HE 20.7008:N84/2

ORIGINAL

COMPLETED
47

PB82-923401

Guidelines for the Prevention and Control of

NOSOCOMIAL INFECTIONS

U.S. DEPARTMENT OF HEALTH AND HUMAN SERVICES
PUBLIC HEALTH SERVICE
CENTERS FOR DISEASE CONTROL
ATLANTA, GEORGIA 30333

BEST DOCUMENT AVAILABLE

Centers for Disease Control ..... William H. Foege, M.D., Director
Center for Infectious Diseases ..... Walter R. Dowdle, Ph.D., Director
John V. Bennett, M.D., Assistant Director
Hospital Infections Program ..... Robert W. Haley, M.D., Director
Guidelines Activity ..... Bryan P. Simmons, M.D., Chief
Publications and Graphics Office ..... Frances H. Porcher, M.A., Chief
Gayle P. Lloyd, M.A.

BEST DOCUMENT AVAILABLE

Contents

Preface

Introduction

GUIDELINE FOR PREVENTION OF CATHETER-ASSOCIATED URINARY TRACT INFECTIONS

- Introduction
- Epidemiology
- Control measures
- Recommendations
- References

GUIDELINES FOR HOSPITAL ENVIRONMENTAL CONTROL

Introduction

Antiseptics, Handwashing, and Handwashing Facilities

- Introduction
- Epidemiology
- Control measures
- Recommendations
- References

Cleaning, Disinfection, and Sterilization of Hospital Equipment

- Introduction
- Epidemiology
- Control measures
- Recommendations
- References

Microbiologic Surveillance of the Environment and of Personnel in the Hospital

- Introduction
- Recommendations
- References

Housekeeping Services and Waste Disposal

- Introduction
- Recommendations
- References

Laundry Services

- Introduction
- Recommendations
- References

Intensive Care Units

- Introduction
- Recommendations
- References

Pharmacy

- Introduction
- Recommendations
- References

GUIDELINES FOR PREVENTION OF INTRAVASCULAR INFECTIONS

Guideline for Prevention of Intravenous Therapy-related Infections

- Introduction
- Epidemiology
- Control measures
- Recommendations
- References

Guideline for Prevention of Infections Related to Intravascular Pressure-monitoring Systems

- Introduction
- Epidemiology
- Recommendations
- References

IV

BLANK PAGE

BEST DOCUMENT AVAILABLE

Preface

This manual, *Guidelines for the Prevention and Control of Nosocomial Infections*, is intended for use by hospital personnel who are responsible for infection surveillance and control. The guidelines have been derived from a variety of sources, including studies conducted by the Centers for Disease Control and by others and have undergone extensive review by experts, many of whom are engaged in the daily practice of infection surveillance and control. The manual is assembled in loose-leaf form to allow periodic revisions and additions.

The topics to be included are listed below. Others may be added in the future. As these revisions and additions become available they may be purchased from:

National Technical Information Service
U.S. Department of Commerce
Springfield, Virginia 22161

Proposed Guideline Topics

1. Prevention of Catheter-associated Urinary Tract Infections
2. Environmental Control of Nosocomial Infections
3. Prevention of Intravascular Infections
4. Prevention of Surgical Wound Infections
5. Prevention of Respiratory Tract Infections
6. Isolation Techniques
7. Role of the Microbiology Laboratory in Infection Surveillance and Control Programs
8. Employee Health Services
9. Objectives and Methods of Surveillance

We welcome all comments, suggestions, and criticisms. Please send them to:

Guidelines Activity
Hospital Infections Program
Centers for Disease Control
Atlanta, Georgia 30333

SUBSCRIPTION INFORMATION

Subscription service for the text portion of this product may be ordered from National Technical Information Service for $50 *(United States, Canada and Mexico) as PB82-923401. It is anticipated the product will be completed in approximately two years.

A three-ring binder, prepared especially for this product, may be ordered for $10 *(United States, Canada, and Mexico) as PB82-923450.

*For orders to other destinations, please contact NTIS for price information.

INTRODUCTION

In the United States nosocomial (hospital-associated) infections occur in approximately 5% of the patients admitted to acute-care hospitals *(1)*, prolong hospital stay by several days on average *(2)*, and lead to over a billion dollars a year in extra direct hospital charges *(3)*. The enormity of the problem has led the Centers for Disease Control (CDC) and other institutions to devote substantial resources over the years to the development of methods and programs to prevent and control such infections. CDC became involved in the 1950s when its medical epidemiologists began to investigate outbreaks of infections in hospitals; in the 1960s more systematic studies of the problem were undertaken *(4,5)*. These early experiences indicated to investigators the importance of adequate knowledge of preexisting infection problems to the efficient detection and control of nosocomial outbreaks and led to the formulation of recommendations for the establishment of organized infection control programs *(5,6)*. Such programs initially emphasized the importance of surveillance, but as the epidemiology and pathogenesis of infections became better defined, hospitals were encouraged to augment their surveillance activities with prevention and control measures *(7-9)*.

In the early 1970s, there was rapid growth in the number of infection surveillance and control programs as hospitals, increasingly concerned about the problem of nosocomial infections, began to adopt infection surveillance and control measures as recommended by CDC and others *(10)*. Despite the proliferation of scientific literature on nosocomial infections, however, it has become obvious that many of these recommendations, although logical and reasonable, have not been validated by well-designed clinical trials; hospitals, faced with rising costs and limited resources, are forced to choose among many potentially expensive surveillance and control measures, often without sufficient scientific guidance.

CDC's recommendations for surveillance and control are based on information obtained from many sources: epidemic investigations, epidemiologic and laboratory studies, consultation and guidance from hospital-based infection control personnel, and the scientific literature. Over the years, these recommendations have been disseminated through numerous scientific symposia, journals, and CDC publications. Since many of these recommendations were published over time and in different sources, they were not readily available to professionals involved in infection control. Since this probably diminished the impact of these recommendations, the need for a central reference—one that would contain CDC recommendations and be easily accessible to the infection control personnel in hospitals—has become obvious.

This manual is being developed to fill that need. It is intended to update and consolidate recommendations for infection surveillance and control into a single, practical document. This edition contains the first of a series of guidelines, each of which addresses an important topic of infection control. Each guideline begins with a preamble that describes the approaches that have been used or advocated to deal with infection control issues and evaluates, where data exist, their efficacy. The preamble is followed by a group of succinct recommendations. The guidelines are assembled in a loose-leaf notebook to allow the addition of new guidelines as they are developed and revisions as necessary.

Optimally, recommendations should be based on rigorously controlled scientific studies because recommendations of this type have the highest probability of value. There are some recommendations that have not been adequately evaluated by controlled scientific trials, but are based on such inherent logic and broad experience that experts generally agree that they are useful. At the other extreme are recommendations that are of uncertain benefit and may be quite controversial. To address these last 2 types of practices, realizing that hospitals must make decisions in the absence of definitive data, we have sought the advice of working groups composed of non-CDC experts with broad experience in infection control. CDC has endorsed such recommendations if members of the working group have determined that the recommendations are likely to be effective.

To assist infection control staff in critically assessing the value of these recommendations, we developed a ranking scheme that takes into account considerations of scientific validity, applicability, and practicality (Table 1). The last 2 considerations are clearly important since scientifically valid infection control practices that are applicable in one setting (e.g., debilitated patients in tertiary referral centers) might not necessarily be applicable or practical in another (e.g., acutely ill patients in community hospitals). Cost effectiveness, another important consideration, is taken into account in the ranking process when possible, although adequate data are generally lacking. We have ranked each recommendation according to the degree to which it has been

BEST DOCUMENT AVAILABLE

substantiated by scientific data or the strength of the working group's opinion on the effectiveness and practical value of the particular practice. The rankings thus provide additional useful information to hospital officials who must decide on the recommendations (e.g., those in Category II and, especially, Category III) that best suit their hospital's needs and resources.

Finally, the adoption of these recommendations by hospitals does not guarantee that hospital personnel will adhere to them. The reduction of nosocomial-infection risks depends largely on the actual performance of correct patient-care practices. Personnel may be motivated to follow these practices if they are given adequate training, followed by periodic in-service education. Continuous or periodic evaluation of patient-care practices, preferably under the supervision of the infection control staff, might assure continued performance of correct practices.

REFERENCES

1. Bennett JV, Scheckler WE, Maki DG, Brachman PS. Current national patterns—United States. In: Proceedings of the International Conference on Nosocomial Infections. Center for Disease Control, August 3-6, 1970.

2. Haley RW, Schaberg DR, Von Allmen SD, McGowan JE Jr. Estimating the extra charges and prolongation of hospitalization due to nosocomial infections: A comparison of methods. J Infect Dis 1980;141:248-57.

3. Dixon RE. Effect on infections in hosital care. Ann Intern Med 1978;89(Part 2):749-53.

4. Cohen LS, Fekety FR, Cluff LE. Studies of the epidemiology of staphylococcal infection. V. The reporting of hospital-acquired infection. JAMA 1962;180:805-8.

5. Eickhoff TC, Brachman PS, Bennett JV, Brown JF. Surveillance of nosocomial infections in community hospitals. I. Surveillance methods, effectiveness, and initial results. J Infect Dis 1969;120:305-17.

6. Garner JS, Bennett JV, Scheckler WE, Maki DG, Brachman PS. Surveillance of nosocomial infections. In: Proceedings of the International Conference on Nosocomial Infections. Center for Disease Control, August 3-6, 1970.

7. Center for Disease Control. Isolation Techniques for Use in Hospitals. Washington, D.C.: U.S. Government Printing Office, 1970. (DHEW publication no. [CDC] 78-8314).

8. Goldmann DA, Maki DG, Rhame FS, Kaiser AB, Tenney JH, Bennett JV. Guidelines for infection control in intravenous therapy. Ann Intern Med 1973;79:848-50.

9. Stamm WE. Guidelines for the prevention of catheter-associated urinary tract infections. Ann Intern Med 1975;82:386-90.

10. Haley RW, Shachtman RH. The emergence of infection surveillance and control programs in US hospitals: An assessment, 1976. Am J Epidemiol 1980;111:574-91.

Table 1. *RANKING SCHEME FOR RECOMMENDATIONS**

Category I. Strongly Recommended for Adoption

Measures in Category I are strongly supported by well-designed and controlled clinical studies that show effectiveness in reducing the risk of nosocomial infections or are viewed as useful by the majority of experts in the field. Measures in this category are judged to be applicable to the majority of hospitals—regardless of size, patient population, or endemic nosocomial infection rate—and are considered practical to implement.

Category II. Moderately Recommended for Adoption

Measures in Category II are supported by highly suggestive clinical studies or by definitive studies in institutions that might not be representative of other hospitals. Measures that have not been adequately studied, but have a strong theoretical rationale indicating that they might be very effective are included in this category. Category II measures are judged to be practical to implement. They are *not* to be considered a standard of practice for every hospital.

Category III. Weakly Recommended for Adoption

Measures in Category III have been proposed by some investigators, authorities, or organizations, but, to date, lack either supporting data or a strong theoretical rationale. Thus, they might be considered as important issues that require further evaluation, they might be considered by some hospitals for implementation, especially if such hospitals have specific nosocomial infection problems or sufficient resources.

***Recommendations that advise against the adoption of certain measures can be found in the guidelines that follow. These negative recommendations are also ranked into one of the 3 categories depending on the strength of the scientific backing or opinions of the members of the working group. A negative recommendation in Category I means that scientific data or prevailing opinion strongly indicate that the measure not be adopted. A negative recommendation in Category III means that, given the available information, the measure under consideration should probably not be adopted; such a measure, however, requires further evaluation.**

BEST DOCUMENT AVAILABLE

Guideline for Prevention of Catheter-associated Urinary Tract Infections

Written by Edward S. Wong, M.D.
In consultation with Thomas M. Hooton, M.D.

WORKING GROUP
Herbert W. Clegg II, M.D.
Children's Hospital Medical Center
Boston, Massachusetts

Sheila Cram, R.N., M.P.H.
Infection Control Nurse
Director of Department of
Infection Control and Epidemiology
Newton-Wellesley Hospital
Newton, Massachusetts

Jane DeGroot-Kosolcharoen, R.N., M.S.
Infection Control Nurse
William S. Middleton Memorial
Veterans Hospital
Lecturer, University of Wisconsin
School of Nursing
Madison, Wisconsin

Richard Garibaldi, M.D.
Associate Professor of Medicine
Hospital Epidemiologist
University of Utah Medical Center
Salt Lake Veterans Administration
Medical Center
Salt Lake City, Utah

Edward H. Kass, M.D.
William Ellery Channing
Professor of Medicine
Director of Channing Laboratory
Brigham and Women's Hospital
Boston, Massachusetts

Calvin M. Kunin, M.D.
Professor and Chairman
Department of Medicine
Columbus, Ohio

Rosemary Lindan, M.D.
Assistant Professor of Orthopedics
and Medicine
Case Western Reserve School of Medicine
Cuyahoga County Hospital
Cleveland, Ohio

Walter E. Stamm, M.D.
Assistant Professor of Medicine
University of Washington
School of Medicine
Harborview Medical Center
Seattle, Washington

Guideline for Prevention of Catheter-associated Urinary Tract Infections

INTRODUCTION

The urinary tract is the most common site of nosocomial infection, accounting for more than 40% of the total number reported by acute-care hospitals and affecting an estimated 600,000 patients per year *(1)*.

Most of these infections—66% to 86%—follow instrumentation of the urinary tract, mainly urinary catheterization *(2)*. Although not all catheter-associated urinary tract infections can be prevented, it is believed that a large number could be avoided by the proper management of the indwelling catheter. The following recommendations were developed for the care of patients with temporary indwelling urethral catheters. Patients who require chronic indwelling catheters or individuals who can be managed with intermittent catheterization may have different needs. Determination of the optimal catheter care for these and other patients with different drainage systems requires separate evaluation.

EPIDEMIOLOGY

The risk of acquiring a urinary tract infection depends on the method and duration of catheterization, the quality of catheter care, and host susceptibility. Reported infection rates vary widely, ranging from 1%-5% after a single brief catheterization *(3)* to virtually 100% for patients with indwelling urethral catheters draining into an open system for longer than 4 days *(4)*. Adoption of the closed method of urinary drainage has markedly reduced the risk of acquiring a catheter-associated infection, but the risk is still substantial. As recent studies have shown, over 20% of patients catheterized and maintained on closed drainage on busy hospital wards may be expected to become infected *(5,6)*. In these studies, errors in maintaining sterile closed drainage were common and predisposed patients to infection. Host factors which appear to increase the risk of acquiring catheter-associated urinary tract infections include advanced age, debilitation, and the postpartum state *(7,8)*.

Catheter-associated urinary tract infections are generally assumed to be benign. Such infection in otherwise healthy patients is often asymptomatic and is likely to resolve spontaneously with the removal of the catheter. Occasionally, infection persists and leads to such complications as prostatitis, epididymitis, cystitis, pyelonephritis, and gram-negative bacteremia, particularly in high-risk patients *(8)*. The last complication is serious since it is associated with a significant mortality, but fortunately occurs in fewer than 1% of catheterized patients *(9,10)*. The natural history of catheter-associated urinary tract infections has been largely unstudied.

Catheter-associated urinary tract infections are caused by a variety of pathogens, including *Escherichia coli*, *Klebsiella*, *Proteus*, enterococcus, *Pseudomonas*, *Enterobacter*, *Serratia*, and *Candida*. Many of these microorganisms are part of the patient's endogenous bowel flora, but they can also be acquired by cross-contamination from other patients or hospital personnel or by exposure to contaminated solutions or non-sterile equipment *(11,12)*. Urinary tract pathogens such as *Serratia marcescens* and *Pseudomonas cepacia* have special epidemiologic significance. Since these microorganisms do not commonly reside in the gastrointestinal tract, their isolation from catheterized patients suggests acquisition from an exogenous source *(13,14)*.

Whether from endogenous or exogenous sources, infecting microorganisms gain access to the urinary tract by several routes. Microorganisms that inhabit the meatus or distal urethra can be introduced directly into the bladder when the catheter is inserted. Generally, however, low rates of infection have been reported after single brief catheterization *(4)*, suggesting that microorganisms introduced by this method are usually removed from healthy individuals by voiding or by antibacterial mechanisms of the bladder mucosa *(15)*. With indwelling catheters, infecting microorganisms can migrate to the bladder along the outside of the catheter in the periurethral mucous sheath *(16,17)* or along the internal lumen of the catheter after the collection bag or catheter-drainage tube junction has been contaminated *(5,6)*. The importance of intraluminal ascension is suggested by the substantial reduction in infections that has been achieved through the use of the closed urinary drainage system. However, if sterile closed drainage can be maintained, extraluminal migration of microorganisms in the periurethral space becomes a relatively more important pathway of entry into the bladder *(17)*.

CONTROL MEASURES

An estimated 4 million patients are subjected yearly to urinary catheterization and, therefore, are at risk for catheter-associated infection and its related sequelae. One of the most important infection control measures is to limit the use of urinary cathe-

ters to carefully selected patients, thereby reducing the size of the population at risk. Generally, urinary catheterization is indicated 1) to relieve urinary tract obstruction, 2) to permit urinary drainage in patients with neurogenic bladder dysfunction and urinary retention, 3) to aid in urologic surgery or other surgery on contiguous structures, and 4) to obtain accurate measurements of urinary output in critically ill patients. Specifically, urinary catheterization should be discouraged as a means of obtaining urine for culture or certain diagnostic tests such as urinary electrolytes when the patient can voluntarily void or as a substitute for nursing care in the incontinent patient.

In selected populations, other methods of urinary drainage exist as possible alternatives to the use of the indwelling urethral catheter. Condom catheter drainage may be useful for incontinent male patients without outlet obstruction and with an intact voiding reflex. Its use, however, requires meticulous nursing care if local complications such as skin maceration or phimosis are to be avoided. In addition, frequent manipulation of the condom catheter drainage system (e.g., by agitated patients) has been associated with an increased risk of urinary tract infection *(18)*. Another alternative, suprapubic catheter drainage, is most frequently used in patients on urologic or gynecologic services. Although preliminary data on the risk of infection are encouraging *(19,20)*, the benefit of the suprapubic catheter with regard to infection control has not been proven by controlled clinical studies. For certain types of patients with bladder-emptying dysfunction, such as those with spinal cord injuries or children with meningomyelocele, a third alternative, intermittent catheterization, is commonly employed. The "no-touch" method of intermittent catheterization advocated by Guttmann *(21)* is generally reserved for patients hospitalized during the acute phase of their spinal cord injury, while the clean, nonsterile method of Lapides *(22)* is frequently used by ambulatory patients for whom the practice of aseptic catheter insertion is difficult to maintain. As with suprapubic catheterization, however, well-designed clinical trials comparing the efficacy of intermittent catheterization by either method to indwelling catheterization in minimizing the risk of infection are lacking.

For patients who require indwelling urethral catheterization, adherence to the sterile continously closed system of urinary drainage is the cornerstone of infection control. For short-term catheterization, this measure alone can reduce the rate of infection from an inevitable 100% when open drainage is employed to less than 25% *(5)*. All other interventions can be viewed as adjunctive measures since none have proven to be as effective in reducing the frequency of catheter-associated urinary tract infections.

Efforts have been made to improve the design of the closed urinary drainage system by modifying or adding to the basic unit introduced and widely adopted in the 1960s. Two modifications, the addition of a urine sampling port in the drainage tubing and the preconnected catheter/collecting tube system seem to have been logical advances since they discourage or prevent opening the closed system which has been well-documented to predispose patients to infection *(6)*. Other alterations have included the insertion of air vents, drip chambers, and one-way valves that were designed to prevent the reflux of contaminated urine. Although these modifications have some theoretical basis, none have been shown to be effective in reducing the frequency of catheter-associated infections. Additionally, overly complex drainage systems can affect the ease of operation or more easily malfunction *(5)*. These latter factors can influence the acceptance of different systems by hospital personnel and ultimately affect infection control.

Other efforts to reduce the incidence of catheter-associated infections have been directed toward 1) preventing microorganisms at the meatus from entering the bladder and 2) eradicating microorganisms that gain entry into the urinary tract before thay can proliferate *(23)*. Measures directed toward the first objective include aseptic catheter insertion, daily meatal cleansing, and daily application of antimicrobial ointments or solutions. On the basis of recent studies that have shown that catheterized patients colonized at the meatus with gram-negative bacilli or enterococci are at increased risk for subsequent infection *(17,24)*, these measures have some theoretical value and can be expected to delay or prevent the onset of infection. Generally, clinical trials that have attempted to demonstrate their efficacy have not been well designed or did not include the use of the closed system of urinary drainage. However, 2 recent, prospective, controlled studies conducted by the same research group have shown that meatal care as it is currently commonly practiced (either twice-a-day cleansing with povidone-iodine solution followed by povidone-iodine ointment or daily cleansing with soap and water) was ineffective in reducing the frequency of catheter-associated infections in patients on closed urinary drainage *(25,26)*. The value of different regimens (e.g., more frequent application, other concentrations, or other antimicrobial agents) is not known and requires further evaluation.

Infection control measures for purposes of eradicating microorganisms in the urinary tract before they can proliferate and cause infection include irrigation of the bladder and the use of prophylactic sys-

temic antibiotics. In one controlled study, continuous irrigation of the bladder with nonabsorbable antibiotics was associated with frequent interruption of the closed drainage system and did not bring about a reduction in the frequency of catheter-associated infections *(27)*. It is not known, however, whether such irrigation would be effective if the integrity of the closed drainage system could be maintained. Several recent studies have shown that prophylactic systemic antibiotics delay the emergence of catheter-related infection *(6,28)*, but this protective effect was transient and was associated with the selection of antibiotic-resistant microorganisms. Thus, controversy regarding the value of prophylactic systemic antibiotics remains.

When cross-infection is likely to be responsible for the spread of catheter-associated infections, additional measures have been proposed *(29)*. In several outbreaks of nosocomial urinary tract infections, catheterized patients with asymptomatic infections served as unrecognized reservoirs of infecting organisms, and the mechanism of transmission appeared to be carriage on the hands of patient-care personnel *(13,14)*. In these outbreaks, the implementation of control measures to prevent cross-infection, including renewed emphasis on handwashing and spatial separation of catheterized patients, particularly infected from uninfected ones, effectively ended the outbreak. In the absence of epidemic spread or frequent cross-infection, spatial separation of catheterized patients is probably less effective in controlling catheter-associated infections.

Regular bacteriologic monitoring of catheterized patients has been advocated to ensure early diagnosis and treatment of urinary tract infections *(8)*. Its possible value as an infection measure lies in its potential usefulness in detecting and initiating treatment of clinically inapparent infections, which may serve as reservoirs of hospital pathogens, and thus, reducing the likelihood of cross-infection. However, the potential benefit of bacteriologic monitoring for such a purpose has not been adequately investigated.

RECOMMENDATIONS

1. Personnel

a. Only persons (e.g., hospital personnel, family members, or patients themselves) who know the correct technique of aseptic insertion and maintenance of the catheter should handle catheters *(5,6,8)*. *Category I*

b. Hospital personnel and others who take care of catheters should be given periodic in-service training stressing the correct techniques and potential complications of urinary catheterization. *Category II*

2. Catheter Use

a. Urinary catheters should be inserted only when necessary and left in place only for as long as necessary. They should not be used solely for the convenience of patient-care personnel. *Category I*

b. For selected patients, other methods of urinary drainage such as condom catheter drainage, suprapubic catheterization, and intermittent urethral catheterization can be useful alternatives to indwelling urethral catheterization *(8,19,21,22)*. *Category III*

3. Handwashing

Handwashing should be done immediately before and after any manipulation of the catheter site or apparatus *(14,30)*. *Category I*

4. Catheter Insertion

a. Catheters should be inserted using aseptic technique and sterile equipment *(8,16,31)*. *Category I*.

b. Gloves, drape, sponges, an appropriate antiseptic solution for periurethral cleaning, and a single-use packet of lubricant jelly should be used for insertion. *Category II*

c. As small a catheter as possible, consistent with good drainage, should be used to minimize urethral trauma *(8)*. *Category II*

d. Indwelling catheters should be properly secured after insertion to prevent movement and urethral traction *(31)*. *Category I*

5. Closed Sterile Drainage

a. A sterile, continuously closed drainage system should be maintained *(5,6,27)*. *Category I*

b. The catheter and drainage tube should not be disconnected unless the catheter must be irrigated (see Irrigation Recommendation 6). *Category I*

c. If breaks in aseptic technique, disconnection, or leakage occur, the collecting system should be replaced using aseptic technique after disinfecting the catheter-tubing junction. *Category III*

6. Irrigation

a. Irrigation should be avoided unless obstruction is anticipated (e.g., as might occur with bleeding after prostatic or bladder surgery); closed continuous irrigation may be used to prevent obstruction. To relieve obstruction due to clots, mucus, or other causes, an intermittent method of irrigation may be used. Continuous irrigation of the bladder with antimicrobials has not proven to be useful *(28)* and should not be performed as a routine infection prevention measure. *Category II*

b. The catheter-tubing junction should be disinfected before disconnection. *Category II*

BEST DOCUMENT AVAILABLE

c. A large-volume sterile syringe and sterile irrigant should be used and then discarded. The person performing irrigation should use aseptic technique. *Category I*

d. If the catheter becomes obstructed and can be kept open only by frequent irrigation, the catheter should be changed if it is likely that the catheter itself is contributing to the obstruction (e.g., formation of concretions). *Category II*

7. Specimen Collection

a. If small volumes of fresh urine are needed for examination, the distal end of the catheter, or preferably the sampling port if present, should be cleansed with a disinfectant, and urine then aspirated with a sterile needle and syringe *(5,8). Category I*

b. Larger volumes of urine for special analyses should be obtained aseptically from the drainage bag. *Category I*

8. Urinary Flow

a. Unobstructed flow should be maintained *(6,8). Category I* (Occasionally, it is necessary to temporarily obstruct the catheter for specimen collection or other medical purposes.)

b. To achieve free flow of urine 1) the catheter and collecting tube should be kept from kinking; 2) the collecting bag should be emptied regularly using a separate collecting container for each patient (the draining spigot and nonsterile collecting container should never come in contact) *(33)*; 3) poorly functioning or obstructed catheters should be irrigated (see Irrigation Recommendation 6) or if necessary, replaced; and 4) collecting bags should always be kept below the level of the bladder. *Category I*

9. Meatal Care

Twice daily cleansing with povidone-iodine solution and daily cleansing with soap and water have been shown in 2 recent studies not to reduce catheter-associated urinary tract infection *(25,26)*. Thus, at this time, daily meatal care with either of these 2 regimens cannot be endorsed. *Category II*

10. Catheter Change Interval

Indwelling catheters should not be changed at arbitrary fixed intervals *(34). Category II*

11. Spatial Separation of Catheterized Patients

To minimize the chances of cross-infection, infected and uninfected patients with indwelling catheters should not share the same room or adjacent beds *(29). Category III*

12. Bacteriologic Monitoring

The value of regular bacteriologic monitoring of catheterized patients as an infection control measure has not been established and is not recommended *(35). Category III*

Summary of Major Recommendations

Category I. Strongly Recommended for Adoption*

Educate personnel in correct techniques of catheter insertion and care.
Catheterize only when necessary.
Emphasize handwashing.
Insert catheter using aseptic technique and sterile equipment.
Secure catheter properly.
Maintain closed sterile drainage.
Obtain urine samples aseptically.
Maintain unobstructed urine flow.

Category II. Moderately Recommended for Adoption

Periodically re-educate personnel in catheter care.
Use smallest suitable bore catheter.
Avoid irrigation unless needed to prevent or relieve obstruction.
Refrain from daily meatal care with either of the regimens discussed in text.
Do not change catheters at arbitrary fixed intervals.

Category III. Weakly Recommended for Adoption

Consider alternative techniques of urinary drainage before using an indwelling urethral catheter.
Replace the collecting system when sterile closed drainage has been violated.
Spatially separate infected and uninfected patients with indwelling catheters.
Avoid routine bacteriologic monitoring.

*Refer to Introduction of manual for full explanation of the ranking scheme for recommendations.

BEST DOCUMENT AVAILABLE

REFERENCES

1. Center for Disease Control. National Nosocomial Infections Study Report. Atlanta: Center for Disease Control, November 1979: 2-14.

2. Martin CM, Bookrajian EN. Bacteriuria prevention after indwelling urinary catheterization. Arch Intern Med 1962; 110:703-11.

3. Turck M, Goffe B, Petersdorf RG. The urethral catheters and urinary tract infection. J Urol 1962;88:834-7.

4. Kass EH. Asymptomatic infections of the urinary tract. Trans Assoc Am Physicians 1956;69:56-63.

5. Kunin CM, McCormack RC. Prevention of catheter-induced urinary tract infections by sterile closed drainage. N Engl J Med 1966;274:1155-62.

6. Garibaldi RA, Burke JP, Dickman ML, Smith CB. Factors predisposing to bacteriuria during indwelling urethral catheterization. N Engl J Med 1974;291:215-8.

7. Brumfitt W, Davies BL, Rosser E. The urethral catheter as a cause of urinary tract infection in pregnancy and puerperium. Lancet 1961;2:1059-61.

8. Kunin CM. Detection, prevention, and management of urinary tract infections. 3rd ed. Philadelphia: Lea and Febiger, 1979.

9. Steere AC, Stamm WE, Martin SM, Bennett JV. Gram-negative rod bacteremia. In: Bennett JV, Brachman PS, eds. Hospital infections. Boston: Little, Brown and Company, 1979:507-18.

10. Kreger BE, Craven DE, McCabe WR. Gram-negative bacteremia IV. Re-evaluation of clinical features and treatment in 512 patients. Am J Med 1980;68:344-55.

11. Selden R, Lee S, Wang WLL, et al. Nosocomial *Klebsiella* infections: intestinal colonization as a reservoir. Ann Intern Med 1971;74:657-64.

12. McLeod JW. The hospital urine bottle and bedpan as reservoirs of infection by *Pseudomonas*. Lancet 1958;1:394-5.

13. Maki DG, Hennekens CH, Bennett JV, et al. Nosocomial urinary tract infection with *Serratia marcesens*: an epidemiologic study. 1973. J Infect Dis 128:579-87.

14. Kaslow RA, Lindsey JO, Bisno AL, Price A. Nosocomial infection with highly resistant *Proteus rettgeri*. Report of an epidemic. Am J Epidemiol 1976;104:278-86.

15. Norden CW, Green GM, Kass EH. Antibacterial mechanisms of the urinary bladder. J Clin Invest 1968;47:2689-700.

16. Kass EH, Schneiderman LJ. Entry of bacteria into the urinary tract of patients with inlying catheters. N Engl J Med 1957;256:556-7.

17. Garibaldi RA, Burke JP, Britt MR, Miller WA, Smith CB. Meatal colonization and catheter-associated bacteriuria. N Engl J Med 1980;303:316-8.

18. Hirsh DD, Fainstein V, Musher DM. Do condom catheter collecting systems cause urinary tract infection? JAMA 1979;242:340-1.

19. Hodgkinson CP, Hodari AA. Trocar suprapubic cystostomy for postoperative bladder drainage in the female. J Obstet Gynecol 1966;96:773-83.

20. Marcus RT. Narrow-bore suprapubic bladder drainage in Uganda. Lancet 1967;1:748-50.

21. Guttman L, Frankel H. The value of intermittent catheterization in the early management of traumatic paraplegia and tetraplegia. Paraplegia 1966;4:63-83.

22. Lapides J, Diokno AC, Gould FR, Lowe BS. Further observations on self-catheterization. J Urol 1976;116:169-71.

23. Sanford JP. Hospital-acquired urinary tract infections. Ann Intern Med 1964;60:903-14.

24. Garibaldi RA, Britt MR, Miller WA, Steinmuller P, Burke JP. Evaluation of periurethral colonization as a risk factor for catheter-associated bacteriuria. In: Proceedings of the 16th Interscience Conference on Antimicrobial Agents and Chemotherapy, 1976:142.

25. Britt MR, Burke JP, Miller WA, Steinmiller P, Garibaldi RA. The non-effectiveness of daily meatal care in the prevention of catheter-associated bacteriuria. In: Proceedings of the 16th Interscience Conference on Antimicrobial Agents and Chemotherapy, 1976:141.

26. Burke JP, Garibaldi RA, Britt MR, Jacobson JA, Conti M, Alling DW. Prevention of catheter-associated urinary tract infections. Efficacy of daily meatal care regimens. In: Proceedings of the 2nd International Conference on Nosocomial Infections, Atlanta, August 4-8, 1980. Am J Med 1981;70:655-8.

27. Warren JW, Platt R, Thomas KJ, Rosner B, Kass EH. Antibiotic irrigation and catheter-associated urinary tract infections. N Engl J Med 1978;299:570-73.

28. Britt MR, Garibaldi RA, Miller WA, Hebertson RM, Burke JP. Antimicrobial prophylaxis for catheter-associated bacteriuria. Antimicrob Agents Chemother 1977;11:240-3.

29. Maki DG, Hennekens CH, Bennett JV. Prevention of catheter-associated urinary tract infection, an additional measure. JAMA 1972;221:1270-1.

30. Steere AC, Mallison GF. Handwashing practices for the prevention of nosocomial infections. Ann Intern Med 1975;83:683-90.

31. Desautels RF, Walter CW, Graves RC, et al. Technical advances in the prevention of urinary tract infection. J Urol 1962;87:487-90.

32. Viant AC, Linton KB, Gillespie WA. Improved method for preventing movement of indwelling catheters in female patients. Lancet 1971;1:736-7.

33. Marrie TJ, Major H, Gurwith M, et al. Prolonged outbreak of nosocomial urinary tract infection with a single strain of *Pseudomonas aeruginosa*. Can Med J 1978;119:593-6.

34. Stamm WE. Guidelines for the prevention of catheter-associated urinary tract infections. Ann Intern Med 1975;82:386-90.

35. Mooney BS, Garibaldi RA, Britt MR. Natural history of catheter-associated bacteriuria (colonization, infection, bacteremia): implication for protection. In: Proceedings of the 11th International Congress of Chemotherapy and the 19th Interscience Conference on Antimicrobial Agents and Chemotherapy, Boston, October 8-12, 1979. Washington, D.C.: American Society of Microbiology, 1980:1083-5.

BEST DOCUMENT AVAILABLE

Guidelines for Hospital Environmental Control

Written by Bryan P. Simmons, M.D.
In consultation with Thomas M. Hooton, M.D., and George F. Mallison, M.P.H.

WORKING GROUP
Edward J. Bertz
Director, Division of Health Facilities and Standards
American Hospital Association
Chicago, Illinois

Mary K. Bruch
Executive Secretary/Microbiologist
Division of Anti-Infective Drug Products
Bureau of Drugs
Food and Drug Administration
Rockville, Maryland

Sue Crow, R.N., M.S.N.
Infection Control Nurse
Louisiana State University Medical Center Hospital
Shreveport, Louisiana

William E. Scheckler, M.D.
Chairman
Department of Family Medicine and Practice
University of Wisconsin
Madison, Wisconsin

Harold Laufman, M.D., Ph.D.
Emeritus Professor of Surgery
Albert Einstein College of Medicine
Emeritus Director of Institute for Surgical Studies
Montefiore Hospital and Medical Center
New York, New York

Janet K. Schultz, R.N., M.S.N.
Assistant Director of Education for Operating Room Consultation
Association of Operating Room Nurses, Inc.
Denver, Colorado

Earle H. Spaulding, Ph.D.
Professor Emeritus and Former Chairman
Department of Microbiology and Immunology
Temple University Health Sciences Center
Philadelphia, Pennsylvania

Richard P. Wenzel, M.D.
Hospital Epidemiologist
Associate Professor of Medicine
University of Virginia Medical Center
Charlottesville, Virginia

BEST DOCUMENT AVAILABLE

INTRODUCTION

Patient-care practices, which usually involve direct personal contact between patients and staff, are the most important factors influencing preventable nosocomial infections. However, the patients and staff—the animate environment—are in constant contact with the inanimate environment, and thus, these 2 environments can and do exchange microorganisms. Further, microorganisms that contaminate the inanimate environment have been associated with sporadic cases of infection and a number of outbreaks. These organisms may be transmitted to patients and staff by a variety of means, direct contact or indirect contact by vehicle, air, or vector.

The inanimate environment must be given proper attention without under- or over-estimating its contribution to infections. Too often, unnecessary attempts are made to disinfect or even sterilize parts of the environment that have rarely been epidemiologically associated with disease. Understanding the role of the environment in disease transmission allows a hospital to focus its personnel and financial resources on areas of most importance.

These guidelines consist of several sections, each describing a different aspect of the hospital's inanimate environment and followed by recommendations. The recommendations were chosen primarily because of their acknowledged importance to infection control, but other factors, such as the feasibility of implementing them and their potential costs to the hospitals, were considered. Many recommendations are intended to reduce expensive "infection control" practices that are not likely to prevent infections.

Reducing the risk of infection caused by environmental contamination depends on adhering to tested protocols for cleaning and processing environmental objects. Personnel may be better motivated to follow these and other guidelines if they are given adequate training after hiring, periodic in-service education, and suitable supervision to assure continued performance.

GENERAL BIBLIOGRAPHY

American Academy of Pediatrics. Standards and recommendations for hospital care of newborn infants. 6th ed. Evanston, Ill.: American Academy of Pediatrics, 1977.

American College of Surgeons Committee on Control of Surgical Infections. Manual on control of infection in surgical patients. Philadelphia: JB Lippincott, 1976.

American Hospital Association. Infection control in the hospital. 4th ed. Chicago: American Hospital Association, 1979.

Bennett JV, Brachman PS, eds. Hospital infections. Boston: Little, Brown and Company, 1979.

Center for Disease Control. Isolation techniques for use in hospitals. 2nd ed. Washington, D.C.: U.S. Government Printing Office, 1975. (DHEW publication no. [CDC] 78-8314).

Cundy KR, Ball W, eds. Infection control in health care facilities. Baltimore: University Park Press, 1977.

Lennette EH, Balows A, Hausler WJ, Truant JP. Manual of clinical microbiology. 3rd ed. Washington, D.C.: American Society for Microbiology, 1980.

U.S. Department of Health, Education and Welfare. Minimum requirements of construction and equipment for hospitals and medical facilities. Washington, D.C.: Government Printing Office, 1979. (DHEW publication no. [HRA] 79-14500).

Antiseptics, Handwashing, and Handwashing Facilities

INTRODUCTION

Handwashing is generally considered the most important single procedure for preventing nosocomial infections. A variety of agents for handwashing—from soap and water to antiseptics—is available in most hospitals. Soap and detergents suspend easily removable microorganisms (and soil) allowing them to be washed off. Antiseptics control or kill microorganisms contaminating skin and other superficial tissues and are sometimes composed of the same chemicals that are used for disinfection of inanimate objects. Although antiseptics and other handwashing agents do not *sterilize* the skin, they can reduce microbial contamination depending on the amount and type of contamination, the agent used, the length of exposure to the agent, the presence of residual activity, and the handwashing technique followed.

In this guideline we define an antiseptic as a product with antimicrobial activity that is designed for use on skin or other superficial tissues. The U.S. Food and Drug Administration (FDA) reserves the term "antiseptic" for a product with antimicrobial activity that has been shown to prevent skin infections in a controlled clinical trial; further, FDA defines antimicrobial-containing products by their intended use, e.g., surgical hand scrub and healthcare personnel handwash.

EPIDEMIOLOGY

Skin flora can be categorized as either resident or transient; resident flora survive and multiply on the skin and can be repeatedly cultured, while transient flora usually survive on the skin less than 24 hours and can be removed quickly and effectively by handwashing. Most resident flora are found on superficial skin layers, but about 10%-20% can inhabit deep layers *(1,2)*. Those in the deep layers cannot be removed by detergents but can be killed or inhibited by antiseptics. They are usually of low virulence and implicated in infections, other than skin infections, only when surgery or other invasive procedures allow them to enter deep tissues or when a patient is highly immunocompromised. Transient skin flora, often found on the hands of hospital personnel, can be pathogens acquired from colonized or infected patients and are frequently implicated in nosocomial infections.

Hospital personnel are believed to be the mode of transmission for most preventable nosocomial infections, and, in many outbreaks, hands of personnel have been identified as the probable means of cross-infection. Handwashing has been shown to eliminate or markedly reduce hand carriage of pathogenic organisms, most of which are transient flora. Thus, it is an important means of preventing patients' exposure to pathogens that have already colonized or infected other patients.

CONTROL MEASURES

The indications for and the ideal frequency of handwashing are generally not known because of the lack of well-controlled studies. The indications for handwashing probably depend on the type, intensity, and duration of patient contact; listing all circumstances that require handwashing would be a lengthy and arbitrary task. Generally, superficial contact with a source not suspected of being contaminated, such as handshaking with most patients, does not require handwashing. In contrast, prolonged and intense contact with any patient should probably be followed by handwashing. In addition, hospital personnel should always wash their hands *before* certain invasive procedures, *before* and *after* contact with wounds, or *before* caring for particularly susceptible patients such as those considered for protective isolation *(3)* and newborn infants. Moreover, hospital personnel should wash their hands *after* touching a source that is likely to be contaminated with hospital pathogens. Hands should be washed before and might need to be washed after the opening of a closed invasive system, because in this context, entering a closed system can be considered analagous to touching a wound. The circumstances that require handwashing are frequently found in special-care units because patients in these units are often infected, sometimes with virulent or multiply-resistant microorganisms, or they are highly susceptible to infection because of wounds, invasive procedures, or diminished immune function. Personnel in these units should wash their hands between contacts with different patients and often need to wash their hands more than once while caring for one patient. In addition, handwashing should be stressed as an important component of the personal hygiene of all hospital employees, and handwashing should be encouraged when personnel are in doubt about the necessity for doing so. However, it should be recognized that frequent handwashing can be time-consuming and, thus, expensive and can cause dermatitis, especially if antiseptics are used.

The recommended handwashing technique depends on the purpose of the washing. For most handwashing in routine patient care, a vigorous, brief washing with soap under a stream of water is recommended because this removes most transient flora *(4)*. Since liquid-soap containers or bar soaps

BEST DOCUMENT AVAILABLE

placed in a pool of water can become contaminated and might serve as a reservoir of microorganisms, containers should be cleaned regularly and bar soaps should be placed on racks that allow water to drain through *(5)*.

Antiseptics (antimicrobial) handwashing products are recommended when personnel need to reliably eliminate microorganisms from their hands; this occurs when the hands are possibly contaminated with virulent microorganisms or whenever the resident skin flora of personnel are likely to cause disease after patient contact. Thus, antiseptics are recommended for handwashing in isolation rooms, in the care of newborn infants, and before invasive procedures such as surgery *(5,6)*. When personnel want to reliably eliminate microorganims with handwashing, they should also remove hand jewelry and clean their nails especially well because jewelry and nails can harbor microorganisms which are not removed with superficial cleaning. Several surgical preoperative handwashing techniques are described in the American College of Surgeons Manual *(7)*; all are acceptable, and one technique does not appear to be clearly better than the others.

Antiseptics are more irritating to skin than soap and water, and frequent washing with them often results in dry skin and dermatitis; paradoxically, this dermatitis can cause an increase in microbial skin colonization and also discourage frequent handwashing. Emollients added to hand creams should decrease or prevent dermatitis and dry skin, but there have been many reports of contaminated hand creams associated with nosocomial infections *(5)*. Consistent use of emollients on the hands after hospital duty rather than during the work day might be efficacious in preventing dermatitis while reducing the risk of these infections.

Chlorhexidine, hexachlorophene, iodophors, and alcohols are the active antimicrobial ingredients recommended for handwashing (Table 1). Aqueous quaternary ammonium products, such as those containing dilute benzalkonium chloride, are not recommended for use on the skin because in-use contamination and subsequent microbial proliferation have been associated with many outbreaks of infection *(8)* and because safer and more effective

Table 1. Characteristics of Antiseptic (Antimicrobial) Agents

Agent	Activity Against Bacteria			Speed of Killing Sensitive Bacteria	Inactivation by Mucus or Proteins	Comments
	Gram-positive	Gram-negative	*Mycobacterium tuberculosis*			
Alcohols	Good	Good	Good	Fast	Marked	Optimum strength 70%-90%, with added emollients, e.g., glycerine or cetyl alcohol, is less drying; "inactivated" by coagulating proteins which then shelter embedded organisms; good for handwashing, although drying, but poor as a single agent for surgical site preparation.
Hexachlorophene: 3% aqueous	Good	Poor-Fair	None	Slow	Moderate	Provides persistent, cumulative activity after repeated use (washing with alcohol reduces persistent action); can be toxic when absorbed from skin, especially in newborns; good for handwashing but *not* for surgical preparation.
Iodine, in alcohol	Good	Good	Good	Fast	Marked	Causes skin "burns", but this is unusual with 1% tincture, especially if it is removed after several minutes; too irritating for handwashing but excellent for surgical site preparation.
Iodophors	Good	Good	Fair	Fast	Moderate	Less irritating to the skin than iodine; has a persistent effect if it is not rinsed off; good for both handwashing and surgical site preparation.
Chlorhexidine: 4% aqueous	Good	Good	Fair	Intermediate	Minimal	Nonirritating, nontoxic, nonsensitizing; has persistent effect; good for both handwashing and surgical site preparation.

NOTE: Some of these agents, such as iodine or chlorhexidine, are combined with alcohol to form tinctures and are available in the combined formulation.
Antiseptics also have activity against fungi and viruses but their relative activity against these is poorly defined.

Adapted with permission, from *Infection Control in the Hospital*, 4th ed., American Hospital Association, 1979.

antiseptics are available.

Antiseptic agents are also used to prepare the skin at the operative site before surgery or other invasive procedures (Table 2). Immediately before a major operative procedure, the site should be thoroughly scrubbed with a sterile detergent solution to remove superficial flora, soil, and debris which could interfere with antisepsis. Then, application of an antiseptic solution is necessary to kill or inhibit more adherent, deep, resident flora. Any of the agents used for antiseptic handwashing, except hexachlorophene, can be used to prepare the operative site, but tincture of chlorhexidine or iodophors offer several advantages over the others *(7,9,10)*. Both have an excellent spectrum of activity against bacteria. Tincture of chlorhexidine has a rapid onset of activity (due to the alcohol) and persistent antibacterial activity. Iodophors also act rapidly and have persistent activity if they are not wiped off. Tincture of iodine is also an excellent surgical prepping agent but can cause skin burns, although this is unusual with a 1% tincture. Alcohols act rapidly but have no persistent effect after they evaporate.

Hexachlorophene has been used as a preoperative skin prep, but it has poor activity against gram-negative organisms and requires repeated use for maximum effectiveness. Hexachlorophene can be absorbed from the skin and in large amounts might have toxic effects on the central nervous system, particularly in premature infants.

The convenient placement of handwashing facilities, single-use (paper) towels, brushes, and manicure sticks where they are needed may facilitate frequent and appropriate handwashing. Immediate recontamination of washed hands can be avoided by using faucets that can be turned off with a paper towel or means other than the hands (foot pedals, etc.) and designing sinks that do not splash easily with running water. Faucet aerators decrease splashing of water in sinks, but they can become contaminated with water-adapted gram-negative bacteria; if aerators are used, periodic removal and cleaning should reduce contamination, at least temporarily.

As an additional precaution, gloves are often worn when the risk of transmitting infection by patient contact is high. Gloves are primarily used by personnel to protect patients from microorganisms that are not removed by handwashing. However, they also protect personnel from blood and secretions that contain virulent microorganisms. When sterile gloves are indicated (Table 2), antiseptic agents are usually recommended for handwashing because gloves are frequently perforated and bacteria multiply rapidly on gloved hands.

RECOMMENDATIONS

1. Handwashing Facilities

Handwashing facilities should be conveniently located for frequent use; preferably, a sink should be located in or just outside every patient room or cubicle with fixed walls. More than one sink per room may be necessary if a large room is used for several patients. *Category II*

2. Handwashing Indications

a. In the absence of a true emergency, personnel should *always* wash their hands 1) *before* performing invasive procedures (Table 2), whether or not sterile gloves are worn; 2) *before* and *after* contact with wounds, whether surgical, traumatic, or associated with an invasive device (e.g., an intravenous cannula entrance wound); 3) *before* contact with particularly susceptible patients; 4) *after* contact with a source that is likely to be contaminated with virulent microorganisms or hospital pathogens, such as an infected patient or an object or device contaminated with secretions or excretions from patients (e.g., a urinary catheter system); and 5) *between* contacts with different patients in special-care units. *Category I*

b. Most routine, brief patient-care activities involving *direct* patient contact other than that discussed in 2a above, e.g., taking a blood pressure or shaking hands, do not require handwashing. *Category II*

c. Most routine hospital activities involving indirect patient contact, e.g., handing a patient medications, food, or other objects, do not require handwashing. *Category I*

3. Routine Handwashing Technique

When handwashing is indicated during *routine* patient care, a vigorous washing under a stream of water for at least 10 seconds is recommended. *Category I*

4. Handwashing Product

a. A handwashing product that is generally acceptable to personnel is desirable for *routine* patient care. Bar soap, a nonantimicrobial liquid soap, granule soap, or soap-impregnated tissues should be used. *Category I*

b. Chlorhexidine, hexachlorophene, iodophors, and alcohol are the recommended active antimicrobial ingredients in handwashing products. Aqueous quaternary ammonium compounds, e.g., benzalkonium chloride, should not be used routinely as antiseptics on skin or tissues. *Category I*

BEST DOCUMENT AVAILABLE

Table 2. Recommended Agents for Preparing Hands and Cleaning Skin Before[a] Non-Surgical and Surgical Procedures

Procedure	Example	Handwashing	Gloves[b]	Preparation Of Patient's Skin	Comment
NONSURGICAL					
Instruments used in the procedure will come in contact with intact mucous membranes	Bronchoscopy; gastrointestinal endoscopy; and tracheal suction	Soap and water	Recommended.	In general, none is required.	
	Cystoscopy; urinary tract catheterization	Soap and water	Sterile recommended.	Antiseptics should be used to prepare the urethral meatus.	
Insertion of a peripheral intravenous or arterial cannula	Intravenous therapy; arterial pressure monitoring	Soap and water or antiseptic	Not necessary but may be helpful in certain instances.	Antiseptics should be used; a fast-acting one is desirable. Tincture of iodine is preferred, but alcohol is adequate if it is applied liberally and allowed to act for 30 seconds.	Most epidemics of infection associated with arterial pressure-monitoring devices appear to be caused by hospital-associated contamination of components external to the skin, such as transducer heads or domes.
Percutaneous insertion of a central catheter or wire	Hyperalimentation; central venous and capillary wedge pressure monitoring; angiography; cardiac pacemaker insertion	Antiseptic	Sterile recommended.	Antiseptics should be used; a fast-acting one is desirable. Tincture of iodine is preferred. "Defatting" agents, such as acetone, are not recommended.	"Defatting" agents do not appear to decrease infections but can cause skin irritation.
Insertion (and) prompt removal) of a sterile needle in deep tissues or body fluids, usually to obtain specimens or instill therapeutic agents	Spinal tap; thoracentesis; abdominal paracentesis	Soap and water or antiseptic	Sterile recommended.	Antiseptics should be used; a fast-acting one is desirable. Tincture of iodine is preferred.	
SURGICAL					
Insertion of a sterile tube or device through tissue into a normally sterile tissue or fluid	Chest tube insertion, culdoscopy; laparoscopy; peritoneal catheter insertion	Antiseptic	Sterile recommended	Antiseptics should be used. Hair should be clipped with scissors, if hair removal is considered necessary.	
Minor skin surgery	Skin biopsy; suturing of small cuts; lancing boils; mole removal	Soap and water	Sterile recommended	Antiseptics should be used.	Gloves are usually worn for a short time and, thus, antiseptic handwashing is not usually necessary to suppress resident flora for these superficial procedures.
Other procedures (major and minor surgery) that enter tissue below the skin	Hysterectomy, cholecystectomy, herniorrhapy	Antiseptic	Sterile recommended.	Antiseptics should be used after the site has been scrubbed with a detergent. The patient can be shaved immediately before the procedure, although clipping hair or using a depilatory is preferred.	Handwashing before surgical procedures that enter deep tissue is usually prolonged to ensure that all areas that harbor bacteria are adequately cleaned.

[a] Hands should be washed *after* all procedures when microbial contamination of the operator is likely to occur, especially those involving contact with mucous membranes, whether or not gloves are worn. Soap and water is adequate for such handwashing unless a virulent agent (e.g., *Shigella* or hepatitis B virus) is suspected of infecting the patient; then an antiseptic should be used.

[b] Gloves protect the patient and the operator from potentially infectious secretions.

BEST DOCUMENT AVAILABLE

5. Indications for Antiseptic (Antimicrobial) Handwashing Product

a. Antiseptics should be used before surgical operations or similar invasive procedures (Table 2). *Category I*

b. Antiseptics should be used upon leaving the rooms of and as otherwise indicated (see Recommendation 2) during the care of patients in strict, respiratory, or enteric isolation; those with wound and skin precautions *(3)*; or those infected with a multiply-resistant bacteria. *Category I*

c. Antiseptics should be used before entering a newborn intensive-care unit or nursery; chlorhexidine, hexachlorophene, and iodophors are preferred. Soap and water is adequate for handwashing between contact with individual patients in these areas unless antiseptics are otherwise indicated (Table 2). *Category II*

d. Antiseptics should be available for use in special care units. *Category I*

e. Antiseptics that do not require water for use, such as alcohol or hexachlorophene foams or chlorhexidine-alcohol rinse, can be used for handwashing in emergency situations in areas where no sinks are available. *Category III*

6. Products to Prepare Operative Site

Tincture of chlorhexidine, iodophors, and tincture of iodine are the recommended products for preparing a patient's operative (invasive) site. Alcohol, as a single agent, is adequate for preparing a patient's skin only for minor procedures such as inserting an intravenous device (Table 2). Plain soap, aqueous quaternary ammonium compounds, or hexachlorophene are not recommended as single agents for operative site preparation unless a patient's skin is sensitive to all acceptable antimicrobial products. *Category I*

7. Hexachlorophene

a. Hexachlorophene should not be used for routine bathing of infants (in the absence of a staphylococcal outbreak). *Category I*

b. Hexachlorophene should not be used by pregnant women. *Category II*

8. Liquid Soap Containers

All liquid soaps should be removed from dispensers at least monthly, preferably more often, and the container cleaned, dried, and refilled with fresh solution. Disposable containers might be preferable. *Category II*

9. Bar Soap

If bar soap is used, it should be changed frequently (a small bar is preferable) and kept on racks that allow drainage of water. *Category II*

10. Handwashing Brushes

Handwashing brushes should be single-use or should be sterilized between uses. Wrapping to maintain sterility before use is not necessary. *Category II*

REFERENCES

1. Price PB. New studies in surgical bacteriology and surgical technique. JAMA 1938;111:1993-6.

2. Ulrich JA. Techniques of skin samoling for microbial contaminants. Hosp Topics 1965;43:121-23.

3. Center for Disease Control. Isolation techniques for use in hospitals. 2nd ed. Washington, D.C.: U.S. Government Printing Office, 1975. (DHEW publication no. [CDC] 78-8314).

4. Lowbury EJL, Lilly HA, Bull JP. Disinfection of hands: removal of transient organisms. Br Med J 1964;2:230-3.

5. Steere AC, Mallison GF. Handwashing practices for the prevention of nosocomial infections. Ann Intern Med 1975; 83:683-90.

6. American Academy of Pediatrics. Standards and recommendations for hospital care of newborn infants. 6th ed. Evanston, Illinois: AAP, 1977.

7. American College of Surgeons Committee on Control of Surgical Infections. Manual on control of infection in surgical patients. Philadelphia: JB Lippincott, 1976.

8. Dixon RE, Kaslow RA, Mackel DC, et al. Aqueous quaternary ammonium antiseptics and disinfectants: use and misuse. JAMA 1976;236:2415-7.

9. Center for Disease Control. Chlorhexidine gluconate: evaluation as an antiseptic. Reprinted from the National Nosocomial Infections Study Report, Annual Summary 1975. Atlanta: Center for Disease Control.

10. Cruse PJE, Foord R. A five-year prospective study of 23,649 surgical wounds. Arch Surg 1973;107:206-10.

FURTHER READING

Chlorhexidine and other antiseptics. Med Lett 1976; 18:21:85-6.

UNPUBLISHED MATERIAL AVAILABLE FROM CDC

Control of nursery-acquired staphylococcal disease: present status of use of hexachlorophene.

Hexachlorophene solutions: a potential reservoir of gram-negative nosocomial pathogens.

Cleaning, Disinfection, and Sterilization of Hospital Equipment

INTRODUCTION

Cleaning is the physical removal of organic material or soil from objects, and it is usually done by using water with or without detergents. Generally, cleaning is not designed to kill microorganisms but to remove them. Sterilization, on the other hand, has as its goal the complete removal or destruction of all forms of microbial life and is carried out in the hospital with steam under pressure, liquid or gas chemicals, or dry heat. Disinfection describes the intermediate measures between physical cleaning and sterilization and is carried out with pasteurization, ultraviolet radiation, or liquid chemicals. The degree of disinfection accomplished depends upon several factors but principally upon the strength of the agent and the nature of the contamination. Some disinfection procedures are capable of producing sterility if they are continued long enough; when these procedures are continued long enough to kill all but resistant bacterial spores, they are called high-level disinfection processes. Other disinfection procedures kill many viruses and most vegetative bacteria but cannot be relied upon to kill resistant organisms such as tubercle bacillus, spores, or certain viruses; these are called low-level disinfection processes. This guideline deals with the cleaning, disinfection, and sterilization of hospital equipment, a task usually performed by the central services department.

EPIDEMIOLOGY

Parts of the hospital environment can be heavily contaminated with potentially pathogenic organisms, but the objects themselves do not usually cause disease unless they are touched or placed onto the body or unless body fluids flow through them. Thus, contaminated *patient-care* supplies or equipment are the most likely objects of the inanimate environment to cause infection. Ironically, contaminated antiseptics (products designed for use on the skin or other tissue) and low-level disinfectants themselves have also been associated with infections.

CONTROL MEASURES

Since it is neither necessary nor possible to sterilize all environmental objects, hospital policies should provide for cleaning, disinfection, or sterilization as necessary to decrease the risk of infection. Which process is indicated for an object depends on the object's intended use and, sometimes, the type of contamination. Any microorganisms, including bacterial spores, which come in contact with normally sterile tissue can cause infection. Thus, it is "critical" that all objects which will touch normally sterile tissues be sterile. However, intact mucous membranes are generally resistant to infection by common bacterial spores but not by many other organisms such as viruses and tubercle bacilli; it is "less critical" that objects touching mucous membranes be sterile, although these objects require a disinfection process that kills all but resistant bacterial spores. Intact skin acts as an effective barrier to all but the most virulent organisms, and it is "not critical" that objects which will touch only intact skin be sterile.

Objects potentially contaminated with virulent organisms, such as hepatitis viruses, *Shigella*, or multiply-resistant gram-negative bacilli, may require disinfection even if their use would normally dictate only cleaning. Tubercle bacilli and polio-, coxsackie-, echo-, and rhino-viruses are resistant to most germicidal agents and require high-level disinfection if they are to be reliably eliminated from reusable objects. Bacterial spores are the most resistant and require a sterilization procedure to be reliably eliminated.

Hospitals should perform most cleaning, disinfection, and sterilization of reusable patient-care objects in a central services department to maintain high levels of quality control. In addition, central processing can save money and free patient-care personnel from such activities. However, some hospitals are able to maintain high levels of quality control in closely monitored processing areas other than the central services department, such as an area adjacent to the operating rooms and in the respiratory therapy department.

Reusable objects must be thoroughly cleaned before processing because organic material (e.g., blood and proteins) inactivate disinfectants and protect microorganisms from disinfection and sterilization. Generally, these objects should be cleaned in the central services department. They should not be precleaned in patient-care areas because such precleaning is inefficient and because cleaning by hand can lead to injury and increased exposure to hepatitis virus. However, some objects that are heavily soiled, such as used bed pans, can benefit from precleaning. Before being sent to central services, objects contaminated with infectious materials or objects from patients in certain types of isolation (1) should be wrapped in impervious plastic and marked "contaminated" in order to decrease exposure of personnel to highly infectious microorganisms. These should be handled with gloves and "decontaminated" before or during cleaning, that is, they should be exposed to a disinfectant or disin-

fecting procedure to render them safe to handle. Moreover, objects exposed to patients in strict isolation that need to be processed in central services should be decontaminated in the room if they cannot otherwise be safely transported *(1,2)*.

For many "noncritical" reusable objects in hospitals such as heat lamps, crutches, wheelchairs, bedboards, and bedside utensils, central processing can consist only of 1) high-temperature washing or hand-scrubbing with a detergent or a disinfectant-detergent combination, 2) rinsing, and 3) thorough drying. Many such objects are not easily moved and can be cleaned where they are used. (A discussion of cleaning that is not done in the central services department or other closely monitored areas is included in the Housekeeping Guideline. Many other objects, including most surgical instruments, can be cleaned by a mechanical washer-sterilizer. However, some delicate instruments might have to be carefully cleaned by hand. Once they are cleaned and inspected, objects requiring sterilization or disinfection are ready for further processing.

Use of pressurized steam in "autoclaves" is the most inexpensive and effective method for sterilization of most objects. Steam sterilization is unsuitable, however, for processing of plastics with low melting points, powders, or anhydrous oils. Residual air pockets can interfere with sterilization in autoclaves, thus, upright containers or steam-impervious wrappers should not be used. After cleaning and before sterilization, objects that will not be used immediately should be wrapped for storage. Sterility can be maintained in storage for various lengths of time depending mainly on the type of wrapping material and the conditions of storage. An item that has been sterilized might not be sterile at the time of use if its safe storage time has been exceeded or if its package has been wet or damaged.

Several methods have been developed to monitor steam sterilization processes, although steam autoclaves are highly reliable if they are used properly. Each sterilizer should be operated according to the manufacturer's instructions. The highest temperature that is reached during sterilization and the length of time that this temperature was maintained should be recorded and checked for adequacy; this check is the most important means of assuring sterility. In addition, heat- and steam-sensitive indicators should be used on the outside of each object. These indicators do not reliably indicate sterility, but they do show that an item has not accidently bypassed a sterilization process. As an additional precaution, a large pack might have an indicator both on the outside and the inside to verify that steam has penetrated the pack. Checks of steam sterilization should be carried out at least once a week using commercial preparations of spores of *Bacillus stearothermophilus* (an organism whose spores are particularly heat resistant, thus assuring a wide margin of safety). If a sterilizer is working properly and used appropriately, the spores are usually killed. A single positive spore test (spores not killed) does not necessarily indicate that objects processed in the same sterilizer are not sterile. It does suggest that the sterilizer should be rechecked for proper temperature, pressure, and use and that the test be repeated. To guarantee a wide margin of safety, spore tests should be done with each load of implantable devices, e.g., implantable orthopedic devices, and these objects should not be released for use until the spore test is negative; packs containing implantable devices need to be clearly labeled so that they can be specially processed. Spore testing should be considered as just one of several methods of assuring adequate processing of inanimate objects in the hospital environment (Table 1).

Because ethylene oxide gas sterilization is a more complex and expensive process than steam sterilization, it is usually restricted to objects that might be damaged by heat or moisture. Before sterilization, objects also need to be cleaned and wrapped. Chemically sensitive indicators should be used with each package to show that it has been exposed to the gas sterilization process. Gas sterilizers should be checked at least once a week with commercial preparations of spores of *Bacillus subtilis* (a resistant spore). All exhaust from gas sterilizers and aerators for gas sterilization should be vented directly to the outdoors because the gas is toxic. All objects processed by gas sterilization need special aeration according to manufacturer's recommendations to remove toxic residues of ethylene oxide.

Liquid chemical disinfection can be effective, but it is time-consuming and expensive. Several chemical solutions are available (Table 2) for high-level disinfection. Solutions containing activated 2% glutaraldehyde can achieve high-level disinfection in 30 minutes and sterilization in 10 hours if the objects are cleaned until they are relatively free of organic matter. Gloves should be worn when using chemical disinfectants to prevent skin reactions, especially rashes. A new formulation, 6% stabilized hydrogen peroxide, does not appear to cause significant skin rashes and can achieve high-level disinfection in 30 minutes and sterilization in 6 hours. Objects disinfected with liquid chemicals must be rinsed in *sterile* water (or water containing at least 10 mg/liter free residual chlorine, e.g., a fresh 1:5000 dilution of a household bleach that is 5.25% hypochlorite solution) to remove possibly toxic or irritating residues. Afterwards, the objects should be handled using sterile gloves and towels and stored in protective wrappers to prevent recontamination.

BEST DOCUMENT AVAILABLE

Table 1. Methods of Assuring Adequate Processing of Inanimate Objects in the Hospital Environment

Object and Classification	Example	Method	Comment
PATIENT-CARE OBJECTS			
Sterility is *critical*			
Sterilized in the hospital, reusable and single use items	Surgical instruments and devices; angiography catheters	Use before maximum safe storage time. Inspect package for integrity and for exposure of sterility indicator before use. Follow manufacturer's instructions for each sterilizer or use recommended protocol. Test sterilizers to find out whether they can kill resistant commercial spores.	Sterilization processes are designed to have a wide margin of safety. If spores are not killed, the sterilizer should be checked for proper use and function; if spore tests remain positive, discontinue use of the sterilizer and have it serviced.
Purchased as sterile	Intravenous fluids; needles; syringes	Use before expiration date if one is given. Inspect package for integrity before use. Culture only if clinical circumstances suggest infection related to use of the item	Notify the U.S. Food and Drug Administration if factory-related (intrinsic) contamination is suspected.
Sterility is *less critical*, but should be free of most vegetative bacteria			
Usually disinfected rather than sterilized in the hospital	Respiratory therapy equipment and instruments for gastrointestinal endoscopy that will touch mucous membranes	Sterilize if possible; if not, follow a protocol for high-level liquid chemical disinfection or wet pasteurization. Perform routine culturing of respiratory therapy and endoscopy equipment	These devices come in contact with mucous membranes. Resistant spores can remain after liquid chemical disinfection, but these are not usually pathogenic. Routine cultures positive for vegetative bacteria should prompt a review of the cleaning/disinfection procedures.
Purchased	Water, including water for hemodialysis	Use an adequately treated source of hospital water. Store fluids with proper chlorination to avoid microbial proliferation. Perform routine culturing of hemodialysis water.	The risk of disease appears to be related to the number of organisms present (unless virulent organisms are present). Water for hemodialysis may require further processing, e.g., deionization.
Sterility is *not critical* and can be expected to be contaminated with some bacteria	Bedpans, crutches, bed rails, water glasses, linens, food utensils, EKG leads, bedside tables, radiology suites, hemodialysis centers	Follow a protocol for cleaning (use a disinfectant or disinfecting process)	These items will not usually come in contact with open skin or mucous membranes.
NON-PATIENT-CARE OBJECTS[a]			
***Likely to be contaminated* with virulent microorganisms**	Laboratories handling patient specimens[b]	Follow a protocol for cleaning (use a disinfectant or disinfecting process).	Areas handling blood or microbiologic specimens are most important.
***Unlikely to be contaminated* with virulent microorganisms**	Areas not involved in patient care: offices, storage areas	Perform routine cleaning.	Cleaning is aimed mainly at improving the appearance of and providing a proper atmosphere in which to work.

[a] Adequate processing of non-patient-care objects is primarily aimed at protecting personnel and others who come in contact with these objects; sterility is *not critical*.

[b] For disposal of specimens from patients, see Guidelines for Hospital Environmental Control: Housekeeping Services and Waste Disposal or *Isolation Techniques for Use in Hospitals* when applicable.

BEST DOCUMENT AVAILABLE

RECOMMENDATIONS

1. General Operation of the Central Services Department

a. No disposable object designed for sterile, single use should be resterilized. *Category I*

b. Any object which should be sterile should not be used if its sterility is seriously questioned, e.g., its package is torn or its expiration date is exceeded. *Category I*

c. Central services should operate to assure that soiled objects do not contaminate those that are clean. Thus, all contaminated objects should be received and decontaminated in one area and cleaned, disinfected, or sterilized elsewhere. These areas should be separate from those used to receive or store new, clean, or sterile objects. *Category I*

2. Decontamination and Cleaning

All objects to be disinfected or sterilized should first be decontaminated, if necessary, and then thoroughly cleaned to remove all blood, tissue, food, and other residue. *Category I*

3. Indications for Sterilization or High-level Disinfection

a. Patient-care equipment that enters normally sterile tissue or the vascular system, or through which blood flows, should be sterile. *Category I*

b. Laparoscopes and other scopes that enter the peritoneal cavity should be subjected to a sterilization procedure before each use; if this is not feasible, they should receive high-level disinfection. *Category I*

c. Endoscopes and respiratory therapy equipment that touch mucous membranes should be subjected to a sterilization procedure before each use, if this is not feasible, they should receive high-level disinfection. *Category I*

4. Methods of Sterilization and Disinfection

a. Whenever sterilization is required, a steam sterilizer should be used unless the object to be sterilized is damageable by heat, pressure, or moisture or is not otherwise amenable to steam sterilization. *Category I*

b. Many reusable patient-care objects can be disinfected or sterilized by the methods found in Table 2. *Category II*

5. Method of Processing, Depending on Contamination

a. Patient-care equipment contaminated with blood from a patient known or suspected to be infected with hepatitis B virus should be sterilized; if this is not feasible, it should receive high-level disinfection. *Category I*

b. Most environmental surfaces contaminated with blood from a patient known to be infected with hepatitis B virus should be cleaned with a solution of household bleach (e.g., a 1/10 dilution of a household bleach that is 5.25% hypochlorite solution) because such solutions have good activity against this virus. Hypochlorite can corrode metal and should be rinsed off. Other high-level disinfectants may be used if hypochlorite is not acceptable. *Category II*

c. Other patient-care and environmental objects that are potentially contaminated with virulent microorganisms should be processed according to the manual *Isolation Techniques for Use in Hospitals (1). Category I*

6. Storage

Sterile packs should be stored no longer than the safe storage time listed in Table 3. The pack must be considered to be contaminated if the wrap is damaged or has been wet. *Category I*

7. Steam Sterilizers

a. Steam sterilizers should be monitored at least once a week with commercial preparations of spores of *Bacillus stearothermophilus*. *Category II*

b. 1) Every load should be monitored with a spore test if it contains implantable objects. These implantable objects should not be used until the spore test is found to be negative (at 48 hours). *Category II*

2) Implantable objects should not be sterilized by "flash" steam sterilization. *Category I*

8. Ethylene Oxide Sterilizers

a. Ethylene oxide sterilization should be limited to objects that must be sterilized but can be damaged by heat, pressure, or moisture. *Category I*

b. Ethylene oxide sterilizers should be monitored at least once a week with commercial preparations of spores of *Bacillus subtilis*. *Category I*

c. Every load should be monitored if it contains implantable objects. These implantable objects should not be used until the spore test is found to be negative (at 48 hours). *Category II*

9. Dry-Heat Sterilizers

a. Dry-heat sterilizers should be monitored at least once a month with commercial preparations of spores of *Bacillus subtilis* that are intended to monitor dry heat. *Category II*

b. Powders and anhydrous oils that must be sterilized should be sterilized by dry heat. *Category II*

BEST DOCUMENT AVAILABLE

Table 2. Methods of Sterilization and Disinfection

	Sterilization		Disinfection	
			High-level	Low-level
	Will enter tissue or vascular system or blood will flow through		Will come in contact with mucous membranes but not enter tissue or vascular system	Will not come in contact with mucous membranes or skin that is not intact
Object	Procedure	Exposure Time (hrs)	Procedure (Exposure Time ≥ 30 min)[a]	Procedure (Exposure Time ≥ 10 min)
Smooth, hard-surface	A	mfr. rec.	C	J
	B	mfr. rec.	D	L
	C	10	E	M
	D	18	F	N
	E	6	G	P
			H	
			I	
			J	
Rubber tubing and catheters[b]	A	mfr. rec.	C	
	B	mfr. rec.	E	
	E	6	F	
			H	
			I	
Polyethylene tubing and catheters[b,c]	A	mfr. rec.	C	
	B	mfr. rec.	D	
	C	10	E	
	D	18	F	
	E	6	H	
			I	
			J	
Lensed instruments	B	mfr. rec.	C	
	C	10	E	
	E	6		
Thermometers (oral & rectal)[d]	B	mfr. rec.	K	
	C	10		
	D	18		
	E	6		
Hinged instruments	A	mfr. rec.		
	B	mfr. rec.		
	C	10		
	E	6		

Key

A Heat sterilization including steam or hot air (see manufacturer's recommendations)
B Ethylene oxide gas (for time, see manufacturer's recommendations)
C Glutaraldehyde (2% aqueous—suitable data is not available to permit an adequate assessment of the comparative effectiveness of dilutions of 2% glutaraldehyde for high-level disinfection of in-use patient care equipment)
D Formaldehyde (8%)-alcohol (70%) solution (corrosion inhibitor needed if formulated in hospital)
E 6% stabilized hydrogen peroxide (will corrode copper, zinc, and brass)
F Wet pasteurization at 75° C for 30 minutes after detergent cleaning
G Sodium hypochlorite (1000 ppm available chlorine) (will corrode metal instruments)
H Phenolic solutions (3% aqueous solution of concentrate)
I Iodophor (500 ppm available iodine)
J Ethyl or isopropyl alcohol (70%-90%)
K Ethyl alcohol (70%-90%)
L Sodium hypochlorite (100 ppm available chlorine)
M Phenolic germicidal detergent solution (1% aqueous solution of concentrate)
N Iodophor germicidal detergent (100 ppm available iodine)
P Quaternary ammonium germicidal detergent solution (2% aqueous solution of concentrate)

Notes

[a] The recommended exposure time for high-level disinfection, 30 minutes, is primarily based on an unpublished CDC study involving disinfection of used respiratory therapy equipment with 2% glutaraldehyde. This time and concentration may differ from those approved by the Environmental Protection Agency for use on product labels. EPA approved times are based on results from manufacturer's studies that are infrequently verified by EPA.

[b] Tubing must be completely filled for disinfection.

[c] Thermostability should be investigated when indicated.

[d] Do not mix rectal and oral thermometers at any stage of handling or processing.

Modified from Center for Disease Control, *Isolation Techniques for Use in Hospitals*, 2nd ed. Washington, D.C.: U.S. Government Printing Office, 1975. (DHEW publication no. [CDC] 78-8314).

BEST DOCUMENT AVAILABLE

Table 3. Safe Storage Times For Sterile Packs

Wrapping	Duration of Sterility[a]	
	In Closed Cabinet	On Open Shelves
Single-wrapped muslin (two layers)[b]	1 week	2 days
Double-wrapped muslin (each two layers)	7 weeks	3 weeks
Single-wrapped two-way crepe paper (single layer)	At least 8 weeks	3 weeks
Tightly woven untreated pima cotton (single layer) over single-wrapped muslin (two layers)	–	8 weeks
Two-way crepe paper (single layer) over single-wrapped muslin (two layers)	–	10 weeks
Single-wrapped muslin (two layers) sealed in 3 mil polyethylene	–	At least 9 months
Heat-sealed, paper transparent plastic pouches	–	At least 1 year

[a] Sterility was checked daily for the first week of storage and weekly thereafter.
[b] Single-wrapped muslin is not recommended because it is easily penetrated by contamination, especially moist contamination.

Augmented, with permission, from *Hospitals, J. A.H.A.*, by the American Hospital Association, copyright October 16, 1974, Volume 48, no. 20.

10. Positive Spore Tests

a. If spores are not killed in *routine* spore tests, the sterilizer should immediately be checked for proper use and function; objects, other than implantable objects, do not need to be recalled for a positive spore test unless the sterilizer or its use is defective. *Category II*

b. If spore tests remain positive after proper use of the sterilizer is documented, its use should be discontinued and it should be serviced. *Category I*

11. Chemical Indicators

Chemical indicators showing that a package has been through a cycle in a sterilizer should be attached to the outside of each package sterilized. *Category I*

12. Preventive Maintenance

Equipment used for disinfection or sterilization should be scheduled for preventive maintenance routinely, according to the manufacturers' instructions. *Category I*

REFERENCES

1. Center for Disease Control. Isolation techniques for use in hospitals. 2nd ed. Washington, D.C.: U.S. Government Printing Office, 1975. (DHEW publication no. [CDC] 78-8314).

2. Center for Disease Control. Recommendations for initial management of suspected or confirmed cases of Lassa fever. Morbid Mortal Weekly Rep 1980; 28(52 suppl):1S-12S.

FURTHER READING

American Hospital Association American Society for Hospital Central Service Personnel. Guidelines for the hospital central service department. Chicago: American Hospital Association, 1978.

Block SS, ed. Disinfection, sterilization and preservation. 2nd ed. Philadelphia: Lea and Febiger, 1977.

Center for Disease Control. Decontamination of CPR training mannequins. Morbid Mortal Weekly Rep 1978;27:132,138.

Association for the Advancement of Medical Instrumentation. Good hospital practice: steam sterilization and sterility assurance. Arlington, Va.: Association for the Advancement of Medical Instrumentation, 1980.

Perkins JJ. Principles and methods of sterilization in health sciences. 2nd ed. Springfield, Ill.: Charles C. Thomas, 1969.

UNPUBLISHED MATERIAL AVAILABLE FROM CDC

Aseptic handling of thermometers and other equipment for measuring patient temperatures. National Nosocomial Infections Study Report, 1976.

Ethylene oxide sterilization.

Recommendations for disinfection and maintenance of respiratory therapy equipment.

Statement on the reuse of disposable items for patient care.

Microbiologic Surveillance of the Environment and of Personnel in the Hospital

INTRODUCTION

Programs for microbiologic sampling in hospitals have often included culturing of purchased sterile products; air; environmental surfaces in patient areas, including surgical suites, nurseries, and other areas; linens; kitchen equipment; and other parts of the inanimate environment. *Routine* sampling programs are those that are conducted on a regularly scheduled basis, irrespective of the characteristics of nosocomial infections in the institution.

With few exceptions, routine programs are not considered cost-effective or justifiable because the occurrence or prevalence of nosocomial infection has not been related to levels of general microbial contamination of air or of environmental surfaces *(1-4)*. Furthermore, meaningful standards for permissible levels of microbial contamination generally do not exist *(1,2)*. Routine sampling generally produces virtually useless information, fosters a sense of false security, and is an unnecessary expense. Unfortunately, in some hospitals environmental sampling has become independent from and has even been substituted for adequate overall programs of surveillance and control of nosocomial infections. The risk of nosocomial infections related to environmental contamination is more effectively controlled by adherence to tested protocols for processing than by routine sampling. Microbiologic sampling should, therefore, be directed toward evaluation of contamination known to be associated with a risk of nosocomial infection or should be used to investigate specific objects or areas that have been incriminated epidemiologically in its transmission. Such sampling should be seen as one of many methods of assuring adequate processing of inanimate objects in the hospital environment (see Table 1 in Guidelines for Hospital Environmental Control: Cleaning, Disinfection, and Sterilization of Hospital Equipment).

Routine programs for monitoring hospital personnel have included mainly the culturing of nose, throat, or stool samples in an attempt to identify carriers of certain bacterial pathogens. But in the absence of disease transmission with a specific pathogen, such culturing does not meaningfully predict the disseminating carrier. Furthermore, the majority of "carriers" do not appear to cause infections. Thus, the recognition of a disease problem should serve as the indication for cultures of hospital personnel.

Hands of personnel are believed to be the major factor in transmission of nosocomial infections, and handwashing is an important means to prevent transmission; some hospitals routinely culture hands of personnel to encourage better handwashing, but this practice can be expensive, and its efficacy has never been studied.

In order to limit unnecessary culturing of the environment or of personnel, such culturing should be directed and guided by the infection control staff and should follow a written plan. A reasonable sampling protocol should outline the specific objects to be sampled, require immediate review of sampling data, and describe what action will be taken based on sampling results.

RECOMMENDATIONS

1. Routine Microbiologic Sampling

Routine microbiologic surveillance of the inanimate hospital environment or of personnel, except for research purposes, should be limited to the following instances:

a. 1) Steam sterilizers should be monitored at least once a week with commercial preparations of spores of *Bacillus stearothermophilus*. *Category II*

2) Every load should be monitored with a spore test if it contains implantable objects. These implantable objects should not be used until the spore test is found to be negative. *Category II*

b. 1) Ethylene oxide ("gas") sterilizers should be monitored at least once a week with commercial preparations of spores of *Bacillus subtilis*. *Category I*

2) Every load should be monitored if it contains implantable objects. These implantable objects should not be used until the spore test is found to be negative. *Category II*

c. Dry heat sterilizers should be monitored at least once a month with commercial preparations of spores of *Bacillus subtilis* that are intended to monitor dry heat sterilization. *Category II*

d. Infant formulas prepared in the hospital should be checked once a week to see that no more than 25 organisms/ml are present (no virulent bacteria, e.g., *Salmonella* or *Shigella*, should be detectable). If bacterial counts exceed this, the preparation technique is probably faulty and should be corrected *(5)*. *Category III*

BEST DOCUMENT AVAILABLE

e. Endoscopes and respiratory therapy equipment that are processed by high-level disinfection should be cultured by a swab or rinse method periodically, based on continuing evaluation of previous results, but at least several times a year. The criterion for satisfactory disinfection after processing is the lack of vegetative microorganisms. *Category III*

f. Water used to prepare dialysis fluid should be tested at least once a month; it should contain a total viable microbial count not greater than 200 microorganisms/ml. The dialysate at the end of dialysis should also be tested once a month and should contain less than 2,000 microorganisms/ml *(6). Category II*

2. Other Indications for Sampling

a. Sampling of the environment or personnel can and should be an integral part of any epidemiologic investigation if environmental or personnel reservoirs are suspected. Culture media can contain antibiotics or other factors that are shown in a pretest to select the epidemic strain. *Category I*

b. Occasional, limited sampling of the inanimate environment can be done to teach and motivate personnel. *Category III*

3. Supervision of Environmental Sampling

a. All microbiologic sampling of the hospital environment or of personnel should be directed and guided by the infection control staff in light of complete epidemiologic information. *Category I*

b. Such sampling should follow a reasonable plan, and the results of the sampling should be reviewed promptly. *Category II*

4. Sampling for Manufacturer-associated Contamination

Patient-care objects purchased as sterile have occasionally been contaminated, but routine sampling of these items is *not* recommended because of the difficulty and expense of performing adequate sterility testing with low-frequency contamination. (If contamination of a commercial product sold as sterile is suspected, the infection control staff should be notified and the nearest district office of the U.S. Food and Drug Administration should be telephoned immediately.) *Category I*

REFERENCES

1. American Hospital Association Committee on Infections within Hospitals. Statement on microbiologic sampling in the hospital. Hospitals 1974; 48:125-6.

2. American Public Health Association Subcommittee on Microbial Contamination of Surfaces. Environmental microbiologic sampling in the hospital. Health Lab Sci 1975; 12:234-5.

3. Mallison GF. Monitoring of sterility and environmental sampling in programs for control of nosocomial infections. In: Cundy KR, Ball W, eds. Infection control in health care facilities. Baltimore: University Park Press, 1977:23-31.

4. Bennett JV, Brachman PS, eds. Hospital infections. Boston: Little, Brown and Co., 1979.

5. American Academy of Pediatrics. Standards and recommendations for hospital care of newborn infants. 6th ed. Evanston, Ill.: American Academy of Pediatrics, 1977.

6. Favero MS, Peterson NJ. Microbiologic guidelines for hemodialysis systems. Dialysis and Transplantation 1977; 6:34-6.

UNPUBLISHED MATERIAL AVAILABLE FROM CDC

Approach to the staphylococcal nasal carrier.

BEST DOCUMENT AVAILABLE

Housekeeping Services and Waste Disposal

INTRODUCTION

Although the role of microbial contamination of environmental surfaces in transmitting nosocomial infections is probably minor, proper housekeeping can decrease the likelihood that large numbers of microorganisms from such surfaces will come in contact with patients.

Recommended cleaning methods vary with the use of the area to be cleaned, the type of surface, and the amount and type of soiling present. Detailed recommendations for housekeeping in operating rooms, nurseries, and isolation areas have been published *(1-5)*. Although it has not been shown that these areas require any special cleaning, they often house patients who are infected with virulent microorganisms or who are particularly susceptible to infection. Such detailed cleaning procedures are intended to provide a margin of safety and, thus, seem warranted. With any cleaning procedure, one should avoid soiling clean areas in the process of cleaning dirty ones; mop heads, cleaning cloths, and cleaning solutions should be changed when they become obviously dirty; and cleaning methods and machines that resuspend dust from surfaces should be avoided, particularly in patient-care areas.

Any hospital-grade disinfectant-detergent registered by the federal Environmental Protection Agency (EPA) can be used for surface cleaning, but physical removal of microorganisms by scrubbing, that is, using "elbow grease," is thought to be more important than any antimicrobial effect of the cleaning agent used *(6)*. Therefore, cost and acceptability by housekeepers can be the main criteria for selecting any such registered agent. However, if used to clean nurseries, disinfectants whose main active ingredient is phenol must be diluted according to instructions on the product label; inadequately diluted solutions have been associated with hyperbilirubinemia in newborns *(7)*.

Disinfectant fogging for control of microbial contamination of air or surfaces is not only ineffective for infection control, it is time-consuming and potentially toxic *(6)*.

The housekeeping service or engineering/maintenance service is usually responsible for devising a plan for disposing of solid wastes that poses minimal risk of disease to patients, employees, and the community. Such plans may vary depending on local ordinances and the availability of acceptable disposal sites. Specific procedures have been recommended *(8)*. Trash (and laundry) chutes, if used, require special design and careful operation because they can give off odor, become fire hazards, and discharge highly contaminated air *(9)*. Infectious material requires special handling and disposal *(5)*.

RECOMMENDATIONS

1. **Frictional Cleaning**
   Because it is the most important factor in environmental cleaning, thorough (elbow-grease) scrubbing should be used for all environmental surfaces that are being cleaned in patient-care areas. *Category I*
2. **Choice of Cleaning Agent for Environmental Surfaces in Patient-Care Areas**
   a. Any hospital-grade disinfectant-detergent registered by the federal EPA can be used. *Category II*
   b. Except for alcohol (70%-90%), antiseptic agents that are intended for use on the skin should not be used for surface cleaning. *Category I*
3. **Disinfectant Fogging**
   Disinfectant fogging should not be done. *Category I*
4. **Routine Cleaning of Horizontal Surfaces**
   In patient-care areas, cleaning of non-carpeted floors and other horizontal surfaces, e.g., bedside tables, should be done daily. *Category II*
5. **Carpeting in Patient-Care Areas**
   a. Carpeting should be vacuumed regularly, cleaned promptly if spills occur, and shampooed (or cleaned with a soil absorbent system) every 3-6 months or whenever its appearance indicates the need for thorough cleaning. *Category II*
   b. Because of problems with cleaning and odors from frequent wetting, carpets should *not* be installed in any area where spillage or heavy soiling is likely, such as operating rooms, obstetrical suites, isolation rooms, emergency rooms, intensive care units, pediatric patient-care areas, kitchens, laboratories, autopsy rooms, bathrooms, and utility rooms. *Category II*
   c. If carpeting is installed in areas where spillage is likely, it should be the indoor-outdoor type, which can be removed for cleaning. *Category II*
   d. Vacuum cleaners should be central or portable units designed to filter discharged air and not resuspend dust from the floor. Many upright vacuum cleaners meet these standards without modification. *Category II*

REFERENCES

1. Mallison GF. Housekeeping in operating suites. AORN J 1975;21:313.

2. American College of Surgeons Committee on Control of Surgical Infections. Manual on control of infections in surgical patients. Philadelphia: JB Lippincott, 1976.

3. Lange K. AORN standards for OR sanitation. AORN J 1975;21:1223-31.

4. American Academy of Pediatrics Committee on Fetus and Newborn. Standards and recommendations for hospital care of newborn infants. 6th ed. Evanston, Ill.: American Academy of Pediatrics, 1977.

5. Center for Disease Control. Isolation techniques for use in hospitals. 2nd ed. Washington, D.C.: U.S. Government Printing Office, 1975.(DHEW publication no. [CDC] 76-8314).

6. Center for Disease Control. Disinfectant fogging, an ineffective measure. National Nosocomial Infection Study Report 1971 (third quarter). Washington D.C.: U.S. Government Printing Office, 1972. (DHEW publication no. [CDC] 72-8149).

7. Wysowski DK, Flynt JW, Goldfield M, et al. Epidemic neonatal hyperbilirubinemia and use of a phenolic disinfectant detergent. Pediatrics 1978;61:165-70.

8. Center for Disease Control. Disposal of solid wastes from hospitals. National Nosocomial Infections Study Report 1974. Washington, D.C.: U.S. Government Printing Office, 1974. (DHEW publication no. [CDC] 74-8257).

9. Hughes HG. Chutes in hospitals. J Can Hosp Assoc 1964;41:56-7.

FURTHER READING

American Hospital Association. Training manual: housekeeping manual for health care facilities. Chicago: American Hospital Association.

Center for Disease Control. Decontamination of CPR training mannequins. Morbid Mortal Weekly Rep 1978;27:132,138.

UNPUBLISHED MATERIAL AVAILABLE FROM CDC

Disinfection of hydrotherapy pools and tanks.

Disposal of solid wastes from hospitals.

Sanitary care and maintenance of ice chests and ice machines.

25 BEST DOCUMENT AVAILABLE

BEST DOCUMENT AVAILABLE

INTRODUCTION

Soiled linens can be a source of large amounts of microbial contamination which may cause infections in hospital patients and personnel, although the risk of infection appears to be low. In addition, improperly processed linens can cause chemical reactions or dermatitis in those who come in contact with them. A hospital's linen service should process soiled linen so that the risk of disease to patients who may be unusually susceptible or to employees who may handle linens is slight. Adequate procedures for collecting, transporting, processing, and storing linens should, therefore, be established.

Washing with hot water and detergent has been shown to result in adequate cleansing of laundry *(1)*; the results of cold-water washing of hospital laundry with modern methods are not known. If needed for other reasons, bleach or ironing may further reduce microbial contamination. Textile softeners added in the final rinse, though of no value in preventing infections, make linen easier to handle and rewash, and they reduce lint.

RECOMMENDATIONS

1. Routine Handling of Soiled Linen

a. Soiled linen should be handled as little as possible and with a minimum of agitation to prevent gross microbial contamination of the air and of persons handling the linen. *Category II*

b. All soiled linen should be bagged or put into carts at the location where used; it should *not* be sorted or prerinsed in patient-care areas. Linen that is saturated with blood or body fluids should be deposited and transported in impervious bags. *Category II*

c. Soiled linen should be removed from patient-care areas at least daily and may need to be removed more frequently, depending on the amount of soiled linen that is generated. *Category II*

2. Handling of Linens Contaminated with Dangerous Microorganisms

a. Linens known to be contaminated with virulent microorganisms, particularly linen from patients in isolation because of infections, should be handled according to recommendations in the manual *Isolation Techniques for Use in Hospitals (2)*; some linens might need to be double-bagged, clearly labeled, and handled with special care. *Category II*

b. An impervious plastic bag should be used for bagging; it can be color-coded and hot-water soluble (some such bags have been known to leak when wet). For double-bagging, the inner bag should be impervious. *Category II*

c. Linens from infected patients in isolation rooms should not be sorted in the laundry before being laundered. *Category II*

3. Transportation of Linen

a. All clean linens should be stored and transported in carts used exclusively for this purpose, or in linen carts that have been decontaminated after being used for soiled laundry. *Category II*

b. If laundry chutes are used, all linens should be bagged. *Category I*

c. Laundry chutes should be located in well-ventilated rooms—not on corridors *(3)*. *Category II*

d. Laundry chutes should be cleaned regularly, e.g., every 6 months, if laundry is not properly bagged. *Category III*

4. Separating Clean from Dirty in the Laundry

In the laundry, dirty linen should be moved from the dirtiest to the cleanest areas as it is being processed; dirty linen should be clearly separated from areas where clean linen is handled. Ventilation air, on the other hand, should flow from the cleanest to the dirtiest areas. All areas should be cleaned on a regular schedule; e.g., some areas that are quickly soiled may require daily cleaning while others may need cleaning twice a year. *Category II*

5. Protecting Personnel Who Sort Laundry

In the laundry, handwashing facilities and protective garb (e.g., gowns, gloves, and masks) should be made available to personnel who sort laundry. *Category II*

6. Hot-Water Washing

Linens should be washed with a detergent in water hotter than 71 °C (160 °F) for 25 minutes since this is an effective method for cleaning and for killing most vegetative bacteria *(1)*. *Category II*

7. Heavily Soiled Items

Heavily soiled items, such as door mats and floor mops, should be laundered separately from linens. *Category II*

8. Sterile Linens

Only those linens used in procedures requiring sterile technique should be sterilized. *Category II*

9. Contract Laundry Services

Laundry services contracted outside the hospital should handle laundry in a manner acceptable to the infection control committee after consideration of the above recommendations. *Category II*

BEST DOCUMENT AVAILABLE

REFERENCES

1. Arnold L. A sanitary study of commercial laundry practices. Am J Public Health 1938;28:839-44.

2. Center for Disease Control. Isolation techniques for use in hospitals. 2nd ed. Washington, D.C.: U.S. Government Printing Office, 1975. (DHEW publication no. [CDC] 76-8314).

3. Hughes HG. Chutes in hospitals. J Can Hosp Assoc 1964;41:56-7.

BEST DOCUMENT AVAILABLE

INTRODUCTION

Patients in intensive care units (ICUs), usually the most seriously ill patients in the hospital, are at increased risk for nosocomial infections for a number of reasons. 1) They often have underlying conditions that compromise their host defenses. 2) They are often exposed to invasive procedures, some of which may be done in emergencies and violate traditional infection control practices. 3) They are often near other highly susceptible or infected patients, a situation which allows ample opportunity for cross-infection since contacts between patients and staff are frequent. Moreover, for a variety of reasons, including the usually high rate of antibiotic use, ICUs often become reservoirs of antibiotic-resistant microorganisms that are difficult to eliminate.

The nature of an ICU makes it an important source of nosocomial infections and thus an important area for infection control. Whenever possible, the design and operation of ICUs should encourage infection control practices, e.g., as might occur if sinks are conveniently placed and patients are in separate cubicles. In addition, because overworked personnel might not take the time necessary for certain aseptic practices, such as handwashing between patient contacts, adequate staffing is important.

Patients in ICUs need to be closely congregated for "intensive care" to be provided with a minimal number of personnel. This consideration must, however, be weighed against the risk of cross-infection that might result from overcrowding.

The American Academy of Pediatrics recommends the wearing of gowns in pediatric ICUs *(1)*, although there is no evidence that the routine use of gowns or other special clothing in pediatric or other ICUs prevents nosocomial infections.

RECOMMENDATIONS

1. Handwashing

Personnel should wash their hands between all patient contacts and when otherwise indicated. Sinks should be provided at convenient locations throughout the ICU and in each of its isolation rooms or alcoves to encourage handwashing. Gloves should be worn for high-risk patient contacts (see Guideline for Hospital Environmental Control: Antiseptics, Handwashing, and Handwashing Facilities, including Table 2 in that guideline). Antiseptics, when indicated, should be easily accessible for handwashing. *Category I*

2. Isolation Facilities

Appropriate isolation facilities should be available in an ICU if infected patients requiring isolation are admitted to these units. *Category I*

3. Spacing Patients

Sufficient space should be provided around each patient for equipment and the passage of personnel to decrease the chance of infection transmission by direct contact *(1,2)*. *Category II*

4. Supervision and In-service Training

Supervision and in-service training should be done in ICUs to ensure that proper handwashing is performed, that disposable equipment is not reused, and that reusable devices are properly cleaned and sterilized. *Category II*

REFERENCES

1. American Academy of Pediatrics Committee on Fetus and Newborn. Standards and recommendations for hospital care of newborn infants, 6th ed. Evanston, Ill.: American Academy of Pediatrics, 1977.

2. U.S. Department of Health, Education and Welfare. Minimum requirements of construction and equipment for hospital and medical facilities. Washington, D.C.: U.S. Government Printing Office, 1979. (DHEW publication no. [HRA] 79-14500).

INTRODUCTION

The hospital's pharmacy is important for infection control because its products are potentially disseminated to all patients. Contamination of medications or other pharmacy products, whether caused by faulty manufacturing, handling, storage, or compounding *(1-3)*, can have disastrous effects on many patients throughout the hospital. Thus, the infection control committee should assist the pharmacy in establishing and assuring aseptic procedures for all compounding and handling of drugs.

Pharmacy personnel have been particularly concerned with preventing bacterial contamination during admixture of parenteral medications, especially hyperalimentation and other intravenous fluids. Several methods for preventing such contamination have been recommended, and although they seem logical, their efficacy and cost-effectiveness have not been determined. They are 1) use of a central pharmacy for all admixture of parenteral medications to ensure better quality control, 2) use of gloves (after handwashing), a gown covering the arms, and a mask to reduce bacterial shedding onto parenterals by the person doing the compounding, 3) use of single-dose vials to prevent cross-contamination, and 4) use of a highly clean area or a "laminar-flow" hood for all admixture. Laminar-flow hoods provide highly filtered and usually sterile air in the working area of the hood. The air has minimal turbulence and is unidirectional, provided objects do not obstruct its flow. Hoods may not provide clean air if they are not properly serviced or if they are used incorrectly; use of proper techniques is important with or without the availability of hoods.

Many of the recommendations that follow are also discussed in the Guideline for Prevention of Intravenous Therapy-related Infections.

RECOMMENDATIONS

1. Location Where Admixture Occurs

a. Parenteral and hyperalimentation fluids should be admixed (compounded) in the pharmacy unless clinical urgency requires admixture in patient-care areas. *Category II*

b. Satellite pharmacy stations, each with its own clean area, might be helpful in some large hospitals. *Category III*

2. Preparation by Personnel for Admixture

a. Personnel should wash their hands before admixing parenterals. *Category I*

b. When admixing large numbers of parenterals at one sitting, personnel should use an antiseptic for handwashing and wear gloves, a gown, and a mask. *Category II*

3. Visual Inspection

All containers of parenteral fluid should be checked for visible turbidity, leaks, cracks, and particulate matter and for the manufacturer's expiration date before admixing and again before distribution. If a problem is found, the fluid should not be used. *Category I*

4. Routine Cultures

Fluid should *not* be routinely cultured, either before or after admixing. *Category I*

5. Labeling

A distinctive supplementary label should be attached to each admixed parenteral stating, as a minimum, the additives and their dosage, the date and time of compounding, the expiration time, and the person who did the compounding. *Category I*

6. Multiple-Use Containers

Single-use (single-dose) containers (vials) should be used for admixture whenever possible. When multiple-use containers intended for intravenous or intramuscular use are opened, they should be marked with the date and time that the container is entered. The product label or package insert should be consulted to determine if refrigeration of the container is necessary. *Category II* (The proper storage temperature is product-specific and is determined by many factors such as stability of ingredients and optimal activity of antibacterial preservatives; bacteria survival in some containers may be enhanced by refrigeration *(4)*. Unless an expiration date is stated on the product label or package insert, it is not known if multiple-use containers, once entered, should be discarded after a specific or arbitrary length of time.)

7. Recording Lot Numbers

The pharmacy should record the appropriate lot numbers of parenteral fluids and other parenteral medications that are dispensed so that they can be traced if contamination is later suspected. *Category III*

8. Refrigeration

a. All admixed fluids should be refrigerated or started within 6 hours of admixture. *Category II*

b. If necessary, admixed parenterals can be stored in the refrigerator for up to 1 week before use provided refrigeration is continuous and begins immediately after admixing.

BEST DOCUMENT AVAILABLE

Other factors, e.g., stability of ingredients, may dictate a shorter storage time.
Category II (This recommendation is intended to prevent waste of parenterals that are admixed for *immediate* use but, unexpectedly, cannot be used.)

9. Antibiotic Usage Programs

In cooperation with the infection control committee, the pharmacy might participate in antibiotic usage programs by providing data on usage patterns *(5). Category III*

10. Laminar-Flow Hood for Admixing Parenterals

a. A laminar-flow hood with horizontal flow should be used for compounding hyperalimentation fluid. *Category II*

b. A laminar-flow hood with horizontal flow should be used for compounding parenteral fluid. *Category II*

11. Proper Use and Maintenance of Laminar-Flow Hoods

If a laminar-flow pharmacy hood is used, the pharmacy should follow the manufacturer's instructions for use and maintenance and should ensure the following:

a. That a minimum number of objects, vials, and other containers are placed in the hood at any time, since they will disrupt airflow. *Category II*

b. That the ultra-high-efficiency filter is tested for integrity and the stated wind velocity is verified at least annually and whenever the hood is moved; a "DOP" smoke-detection system (a test using a standard small-particle aerosol) should be used to test for filter integrity (contact manufacturer of hood for details). *Category II*

c. That all surfaces inside the hood (except filter surfaces) are wiped clean with 70%-90% ethyl or isopropyl alcohol or a disinfectant-detergent solution before each use and several times a day if the hood is in continuous use. *Category II*

REFERENCES

1. Herman LG. A critical evaluation of microbiological hazards associated with pharmacy and the hospital. Am J Hosp Pharm 1970;27:56.

2. Kallings LO. Contamination of therapeutic agents. In: Proceedings of the International Conference on Nosocomial Infections, Atlanta, August 3-6, 1970. Chicago: American Hospital Association, 1971: 241.

3. Maki DG, Rhame FS, Mackel DC, et al. Nationwide epidemic of septicemia caused by contaminated intravenous products. I. Epidemiologic and clinical features. Am J Med 1976;60:471-85.

4. Lehmann CR. Effect of refrigeration on bactericidal activity of four preserved multiple-dose injectable drug products. Am J Hosp Pharm 1977;34:1196-1200.

5. Joint Commission on Accreditation of Hospitals. Accreditation manual for hospitals. 1980 ed. Chicago: the Joint Commission on Accreditation of Hospitals, 1980.

BEST DOCUMENT AVAILABLE

Guidelines for Prevention of Intravascular Infections

Written by Bryan P. Simmons, M.D.
In consultation with Thomas M. Hooton, M.D.,
Edward S. Wong, M.D.,
and James R. Allen, M.D., M.P.H.

WORKING GROUP
Kenneth Barker, Ph.D.
Professor and Head of the
Department of Pharmacy Care Systems
School of Pharmacy
Auburn University
Auburn, Alabama

Rita Colley, R.N.
Clinical Nurse Specialist/
Administrative Director
Hyperalimentation Unit
University of Cincinnati Medical Center
Cincinnati, Ohio

Mary Larkin, R.N.
Intravenous Nurse
Executive Director,
National Intravenous Therapy Association
Belmont, Massachusetts

Michelle M. Maher, R.N.
Clinical Research Nurse—
Total Parenteral Nutrition
National Institutes of Health
Clinical Center Nursing Service
Bethesda, Maryland

Dennis G. Maki, M.D.
Head, Infectious Disease Section
Associate Professor of Medicine
Hospital Epidemiologist
and
Director, Trauma and Life
Support Center
University of Wisconsin Hospitals
Madison, Wisconsin

John E. McGowan, Jr., M.D.
Associate Professor
Department of Medicine
(Infectious Diseases) and
Department of Community Health
Emory University School of Medicine
Hospital Epidemiologist
Grady Memorial Hospital
Atlanta, Georgia

Robert A. Weinstein, M.D.
Hospital Epidemiologist
Michael Reese Hospital
Assistant Professor of Medicine
University of Chicago
Chicago, Illinois

Guideline for Prevention of Intravenous Therapy-related Infections

INTRODUCTION

Intravenous (IV) therapy is an integral part of modern patient care and is given to approximately 30% to 50% of hospitalized patients, especially those who are seriously ill. An IV system offers a ready means of direct access to a patient's vascular system for hemodynamic monitoring and for administering numerous agents that cannot be administered as effectively by other means. Unfortunately, IV systems also provide a potential route for microorganisms to enter the vascular system by bypassing normal skin defense mechanisms; these organisms can cause serious infection if they are allowed to enter and proliferate in the IV cannula (catheter) wound or IV fluid. IV therapy, therefore, is a potential cause of serious illness or death for hospitalized patients.

EPIDEMIOLOGY

IV-related infections consist of those related to microbial contamination of the cannula and the cannula wound (cannula-related) and those related to microbial contamination of the infusate (infusate-related). Most IV-related infections are cannula-related. Cannula-related infections are generally endemic, i.e., not part of an outbreak, while most recognized infusate-related infections are epidemic.

Cannula-related infections, which can occur with or without fever or bacteremia, include purulent thrombophlebitis, cellulitis, and "occult IV site infection." These infections are frequently preceded by or associated with phlebitis (usually characterized by warm, erythematous skin over an indurated or tender vein). Although phlebitis can be caused by infection, it is usually caused by mechanical or chemical irritation from the cannula or infusate. Purulent thrombophlebitis often has the same signs and symptoms as uncomplicated phlebitis but, in addition, is characterized by gross or microscopic evidence of suppuration within the lumen of the vessel; pus sometimes drains spontaneously from the cannula wound, but pressure may be required to express it. This infection is dangerous in that it frequently leads to high-grade bacteremia and often requires surgical removal of the vein for resolution. Cellulitis is characterized by warm, erythematous, and often tender and edematous skin surrounding the site of cannula insertion; pus is rarely detectable. "Occult IV-site infection" is a broadly defined term used to describe infections that characteristically do not produce much (if any) pus or inflammation at the IV site. Early in their course, these infections often have no systemic symptoms or signs. However, because they are the most common cannula-related infections, they probably cause bacteremia more frequently than purulent thrombophlebitis or cellulitis do. Thus, occult IV-site infections pose an important and difficult diagnostic problem; diagnosis may depend on culturing of the cannula.

Factors that influence a patient's risk of acquiring a cannula-related infection include 1) patient susceptibility to infection; for example, certain burn and immunocompromised patients are more susceptible to these infections; 2) the type of cannula used; for example, plastic cannulas are generally associated with a higher rate of infection than steel ("scalp-vein") cannulas; 3) the method of insertion; for example, cannulas inserted by cutdown appear to be associated with a higher rate of infection than those inserted percutaneously; 4) duration of cannulation; for example, peripheral cannulas that have been in continuous use for longer than 48-72 hours are associated with a marked increased rate of infection; and 5) the purpose of the cannula; for example, central cannulas that are used for monitoring central venous pressure (CVP) are associated with comparatively high rates of infection. Because of differences among hospitals with regard to the factors that influence infection risk, reported rates of infection vary considerably. However, the frequency of 2 important complications has been estimated by combining data from a number of hospitals: Purulent thrombophlebitis occurs in about 0.2% and IV-related bacteremia in less than 1% of all patients who receive IV therapy *(1)*.

Staphylococcus aureus, a gram-positive bacterium, is the single microorganism most frequently recovered from cannula-related infections, accounting for about 30% of the pathogens identified; but gram-negative bacteria (as a group) account for about 50% *(1)*. The spectrum of pathogens does not appear to differ with the type of cannula used or hospital service, but the use of antibiotic and antiseptic ointments at the cannula site can change the flora encountered (e.g., from bacteria to fungi) and their antibiotic susceptibility patterns.

Determining whether bacteremia is related to the cannula is frequently difficult, since skin flora, especially *Staphylococcus epidermidis*, predominate when catheter tips are routinely cultured in broth, and the large majority of these organisms are not suspected of causing infection. Maki et al., however, described a semiquantitative method of culturing IV cannulas that appears to distinguish infection from contamination *(2,3)*. High-density colonization of the cannula, as determined by this culture technique, correlates significantly with IV-related bacteremia and local inflammation.

Because frequent culturing is expensive and because most cannulas are not associated with infection, hospital personnel need to know when culturing an IV cannula is most likely to be helpful. In general, culturing should be useful whenever IV-related infection is suspected. Purulent thrombophlebitis or cellulitis at an IV site strongly suggests the presence of an IV-related infection, but culturing the cannula can confirm the diagnosis and isolate the causative organism; any gross pus should be gram-stained and cultured also. Culturing the cannula is also likely to be helpful whenever a patient receiving IV therapy develops fever or bacteremia and no focus of infection is apparent; culturing by the semiquantitative method can detect infection even when there is no visible evidence of infection at the IV site.

Infections related to microbial contamination of infusate occur much less frequently than cannula-related infections, but when they do occur, they are generally epidemic *(4,5)*. Contamination can occur during manufacturing and before use (intrinsic contamination) or during hospital preparation or use (extrinsic contamination) when organisms are introduced during manipulation of the IV bottle, tubing, or devices connected to the tubing (piggyback infusions, stopcocks, etc.). Sepsis from the administration of contaminated infusates may be clinically indistinguishable from sepsis from other causes, but suspicion of this problem should be aroused when 1) signs and symptoms of sepsis appear immediately or shortly after infusion is initiated, especially when they occur in a patient in whom sepsis is unlikely and when there is no apparent focus of bacteremia; 2) antibiotic therapy with appropriate agents fails to resolve the sepsis as long as the infusion continues; 3) the patient rapidly improves when IV therapy is discontinued, with or without the use of antibiotics; or 4) the same organism is isolated from the patient's infusate and blood.

Infections caused by contaminated infusate are almost exclusively due to gram-negative bacilli, primarily members of the tribe Klebsielleae (*Klebsiella*, *Enterobacter*, and *Serratia* species), although *Citrobacter freundii* and *Pseudomonas thomasii* (*cepacia*) have also been implicated in epidemics. These organisms are able to proliferate in glucose-containing IV fluid; most other organisms either die or do not proliferate in such fluid *(4,5)*. Isolation of one of these organisms from the bloodstream of a patient believed to have IV-related infection should suggest contaminated infusate rather than the more frequent cannula-related infection, although either may be responsible. When these organisms have been isolated in epidemics caused by intrinsically contaminated infusate, their antibiotic susceptibility patterns have often been distinct from nosocomial bloodstream pathogens usually encountered in the institution, in that they have not demonstrated the usual multiple antibiotic-resistance patterns *(5)*.

Hyperalimentation fluids, especially those containing crystalline amino acids, inhibit the growth of most microorganisms. However, *Candida* can proliferate in such fluids, albeit slowly *(6)*. Lipid emulsions support rapid growth of a variety of microorganisms *(7)*. Infection control recommendations for hyperalimentation will not be discussed in this guideline.

CONTROL MEASURES

Most control measures dealing with IV therapy are designed to encourage proper use of cannulas since cannula-related infections account for most IV-related infections. One of the first steps in initiating IV therapy is to decide what type of cannula should be used. Steel-needle cannulas have been associated with a lower risk of infection than plastic cannulas; however, the studies that have compared them have not been specifically randomized and controlled for type of cannula. Recently, a randomized, controlled study comparing steel and plastic cannulas showed no difference in the rates of infection related to the 2 types of cannula *(8)*; however, this study excluded some patients at highest risk of infection, those in intensive care units, and was done in a hospital with an IV team that routinely changed cannulas at least every 72 hours. It appears reasonable to conclude from this study that plastic cannulas are safe for routine use in hospitals where all cannulas are changed every 48-72 hours (such as may be done by an IV team). In addition, because plastic cannulas tend to infiltrate or dislodge less frequently than steel cannulas, they are indicated whenever a secure access to the vascular system is needed and even temporary interruption of IV therapy might have serious sequelae. Further, plastic cannulas must be used for cannulation of central veins. Otherwise, stainless steel cannulas are probably safer for IV infusions.

The method and site of cannula insertion also appear to affect the rate of IV-related infections and complications. Insertion by percutaneous puncture rather than by cutdown is associated with a lower risk of infection *(1)*. In adults, upper extremity, subclavian, and jugular sites of cannula insertion are associated with fewer complications, especially thrombophlebitis, than lower extremity sites.

Since most IV-related infections are postulated to result from inward progression of microorganisms contaminating the cannula wound, many control measures are aimed at preventing contamination of the insertion site. These include handwashing by personnel before cannula insertion, preparation of the IV site with an antiseptic, and application of an

antibiotic or antiseptic ointment at the skin-cannula junction at the time of insertion. Topical polyantibiotic ointments appear to reduce infections when used for insertions requiring a cutdown and may be efficacious for other insertions; antiseptic ointments (e.g., povidone iodine ointments) afford little, if any, benefit for most insertions *(9,10)*.

Other measures intended to prevent contamination of the insertion site include use of sterile dressings, acetone, subcutaneous tunneling, and methods of securing the cannula. Covering the IV site with a sterile dressing should reduce contact with contaminated hands and other objects. A recently developed transparent, semipermeable membrane dressing is being promoted for use on wounds, including IV cannula wounds; not enough information is available to evaluate the role of this dressing for cannula wounds. Cleaning the insertion site with acetone has been recommended to remove organic debris that may support bacterial growth, but this practice does not appear to prevent infection and appears to increase skin irritation *(11)*. Inserting the cannula so that it enters the skin at a site away from where it enters the vein, i.e., subcutaneous tunneling, is a technique advocated for cannulas that are inserted into central veins and is intended (but this has not been proven) to block inward migration of microorganisms that contaminate the insertion site *(12,13)*. Securing the IV cannula after insertion may reduce motion that can injure the vein or, possibly, allow organisms to be pushed deeper into the cannula wound.

Regular maintenance and observation of the IV site is directed toward prevention or, at least, early detection of IV-related complications, such as phlebitis, or other signs that may indicate infection. Many experts recommend inspecting the IV site every 24 hours because this seems to be an adequate interval to detect early complications. Visual inspection of the insertion site is probably not necessary if 1) the site is palpated gently through the intact dressing, and no pain or tenderness is present, 2) the patient has no unexplained fever, and 3) peripheral cannulas are removed every 48-72 hours. Removing of peripheral cannulas after 48-72 hours is recommended because of the sharp increase in the rate of infection after this length of time. Recording the date and time of cannula insertion in the medical record or in another easily accessible location has been advocated to remind personnel of when to inspect the site or remove the cannula.

Control of infections related to contaminated infusate depends on 1) adequate manufacturing and quality control processes to ensure sterility, 2) aseptic compounding in hospitals, 3) prompt use of compounded parenterals, and 4) procedures to prevent in-use contamination. Other than reporting suspected intrinsic contamination of IV fluids to the U.S. Food and Drug Administration, individual hospitals generally cannot influence the first condition but can influence the last 3. Compounding of parenterals in the pharmacy rather than in patient-care areas might improve quality control and reduce exposure to hospital pathogens that can contaminate parenterals, especially if pharmacists are trained and experienced. Inspection of the IV container before compounding can detect leaks, cracks, or precipitants that suggest contamination, although it must be recognized that fluid contaminated with more than 10^5 bacteria per milliliter can appear normal and clear. Refrigeration will retard growth of microorganisms in IV fluid, and with hyperalimentation fluid, refrigeration at 4 °C has been shown to prevent growth for at least 7 days *(6)*. However, the effect of refrigeration at temperatures higher than 4 °C, a condition that probably exists in many hospital refrigerators, has not been studied. At room temperature, organisms that grow in IV fluid, mainly those of the tribe Klebsielleae, can proliferate to large numbers in 24 hours; thus, experts usually recommend refrigeration of admixed IV fluids until use and discarding in-use IV fluids after 24 hours.

Administration sets and the fluid flowing through them can become contaminated during in-use manipulations, such as injecting medications and flushing lines, of the IV system. Special ports in the administration set are designed to reduce contamination by providing a means of adding medications without disconnecting a closed IV system. Once contaminated, sets can act as a nidus for microbial proliferation; therefore, routine changing is recommended. However, there is controversy about the proper interval for such changes. A 48-hour interval seems to result in no excess infection when compared with a 24-hour interval *(14,15)*. For administration sets used for hyperalimentation, a 24-hour changing interval is recommended by most authorities, although these sets appear no more prone to contamination than sets used for other types of infusions. In addition, some authorities recommend changing sets immediately after the administration of blood, blood products, or lipid emulsions because these agents greatly increase the spectrum of organisms that grow in IV fluid *(16)*.

Some individuals and institutions have introduced additional measures to reduce IV-related infection. Many hospitals, recognizing that even the best IV-care protocols are not always followed by busy physicians and nurses, have instituted specially trained IV teams to insert IV cannulas and maintain IV cannulas and infusions. These teams, especially those used for hyperalimentation, appear promising in reducing IV-related infections; however, no con-

trolled trial has studied their effectiveness. IV filters have been added to the terminal end of administration sets in some institutions but have not been shown to reduce IV-related infection; they are, however, advocated by some for preventing a variety of noninfection complications of IV therapy.

RECOMMENDATIONS

1. Indications for Use

Intravenous (IV) therapy should be used only for definite therapeutic or diagnostic indications. *Category I*

2. Choice of Cannulas for Peripheral Infusions

a. Plastic cannulas are acceptable for routine peripheral IV infusions if and only if the hospital can be sure that cannulas are replaced every 48-72 hours, such as can be done by an IV team. *Category II*

b. Otherwise, stainless steel cannulas should be used for routine peripheral IV infusions, and plastic cannulas should be reserved for those clinical settings when a secure route for vascular access is imperative. *Category II*

3. Handwashing

a. Hospital personnel should wash their hands before inserting an IV cannula. *Category I*

b. Soap and water is adequate for handwashing for most insertions, but an antiseptic should be used before insertion of central cannulas and cannulas requiring a cutdown. *Category II*

c. Sterile gloves should be worn for insertion of central cannulas and cannulas requiring a cutdown. *Category I*

4. Choice of Site

In adults, the upper extremity (or if necessary, subclavian and jugular sites) should be used in preference to lower extremity sites for IV cannulation. All cannulas inserted into a lower extremity should be changed as soon as a satisfactory site can be established elsewhere. *Category I*

5. Site Preparation

a. The IV site should be scrubbed with an antiseptic prior to venipuncture. *Category I*

b. Tincture of iodine (1%-2%) is preferred, but chlorhexidine, iodophors, or 70% alcohol can be used. The antiseptic should be applied liberally and allowed to remain in contact for at least 30 seconds prior to venipuncture. *Category II*

c. Neither aqueous benzalkonium-like compounds nor hexachlorophene should be used to scrub the IV site. *Category I*

6. Procedures Accompanying Insertion

a. A topical antibiotic or antiseptic ointment should be applied at the IV site immediately after cannula insertion, especially for insertions by cutdown. *Category II*

b. The cannula should be secured to stabilize it at the insertion site. *Category I*

c. A sterile dressing should be applied to cover the insertion site. The dressing, and not tape, should cover the wound unless the tape is sterile. *Category I*

d. The date of insertion should be recorded in a place where it can be easily found. *Category I* (The date may be recorded in the medical record and, if feasible, also on the dressing or tape.)

7. Maintenance of IV Site

a. Patients with intravenous devices should be evaluated at least daily for evidence of cannula-related complications. This evaluation should include gentle palpation of the insertion site through the intact dressing. If the patient has an unexplained fever or there is pain or tenderness at the insertion site, the dressing should be removed and the IV site inspected. *Category I*

b. For peripheral cannulas that must remain in place for prolonged periods, the IV site should be inspected and dressed with a new sterile dressing at 48-72 hours. Thereafter, the site should be inspected and dressed regularly (the optimal frequency of dressing cannula sites in this situation is not known). *Category II*

c. Antibiotic or antiseptic ointment, if used, should be reapplied with each dressing change. *Category II*

8. Replacement of Peripheral Cannula

If prolonged IV therapy with a peripheral cannula (including heparin-lock devices and peripheral cannulas inserted by a cutdown) is indicated, the cannula should be changed and a new cannula inserted every 48-72 hours provided no IV-related complications requiring cannula removal are encountered before this. Cannulas inserted without proper asepsis, for example, those inserted in an emergency, should be replaced at the earliest opportunity. *Category I* (Peripheral cannulas may occasionally have to be used for longer than 48-72 hours if another peripheral site cannot be found.)

9. Special Procedures for Central Cannulas (those whose tips lie in the large central vessels)

a. Central cannulas should be inserted with aseptic technique and sterile equipment. Gloves and drapes are usually required to achieve this objective. *Category I*

b. All central cannulas should be removed when they are no longer medically indicated or if they are strongly suspected of causing sepsis. *Category I*

c. Central cannulas that are inserted through a subclavian or jugular approach, except those used for pressure-monitoring, need not be changed routinely. *Category I* (The proper frequency for changing cannulas that are used for pressure-monitoring, for example, those used to measure pulmonary artery or central venous pressure, is not known.)

d. Central cannulas that are inserted through a peripheral vein should be treated as peripheral cannulas (see appropriate recommendations above). *Category I*

e. For central cannulas that must remain in place for prolonged periods, the insertion site should be inspected and dressed with a new sterile dressing every 48-72 hours. *Category II*

10. Maintenance of Administration Sets

a. IV administration tubing, including "piggyback" tubing, should be routinely changed every 48 hours. *Category I*

b. Tubing used for hyperalimentation should be routinely changed every 24-48 hours. *Category II*

c. Tubing should also be changed after the adminstration of blood, blood products, or lipid emulsions. *Category III*

d. Between changes of components, the IV system should be maintained as a closed system as much as possible. All entries into the tubing, as for administration of medications, should be made through injection ports that are disinfected just before entry. *Category I*

e. Flushing or irrigation of the system to improve flow should be avoided. *Category II*

f. Blood specimens should not be withdrawn through IV tubing except in an emergency or when immediate discontinuation of the cannula and tubing is planned. *Category II*

11. Changing Parts of the IV System for Infection or Phlebitis

a. The entire IV system (cannula, administration set, and fluid) should be changed immediately if purulent thrombophlebitis, cellulitis, or IV-related bacteremia is noted or strongly suspected. *Category I*

b. For phlebitis without signs of infection, the cannula should be changed. *Category I*

12. Culturing for Suspected IV-related Infections

a. If an IV system is to be discontinued because of suspected IV-related infection, such as purulent thrombophlebitis or bacteremia, the skin at the skin-cannula junction should be cleaned with alcohol and the alcohol allowed to dry before cannula removal, and the cannula should be cultured using a semi-quantitative technique *(2,3)*. *Category II*

b. 1) If an IV system is discontinued because of suspected fluid contamination, the fluid should be cultured *(5)* and the implicated bottle saved. *Category I*

2) If an IV system is discontinued because of suspected IV-related bacteremia, the fluid should be cultured. *Category III*

3) If contamination of fluid is confirmed, the implicated bottle and the remaining units of the implicated lot should be saved, and the lot numbers of fluid and additives should be recorded. *Category I*

4) If *intrinsic* contamination (contamination during manufacturing) is suspected, the local health authorities, CDC, and the U.S. Food and Drug Administration should be notified immediately. *Category I*

13. Quality Control During and After Admixture

a. Parenteral and hyperalimentation fluids should be admixed (compounded) in the pharmacy unless clinical urgency requires admixture in patient-care areas (see Guideline for Hospital Environmental Control: Pharmacy). *Category II*

b. Personnel should wash their hands before admixing parenterals. *Category I*

c. All containers of parenteral fluid should be checked for visible turbidity, leaks, cracks, and particulate matter and for the manufacturer's expiration date before admixing and before use. If a problem is found, the fluid should not be used and should be sent to (or remain in) the pharmacy. *Category I*

d. In the pharmacy, a laminar-flow hood should be used for admixing parenteral fluid. *Category II*

e. Single-use (single-dose) containers (vials) should be used for admixture whenever possible. When multiple-use containers intended for intravenous use are opened, they should be marked with the date and time that the container is entered. The product label or package insert should be consulted to determine if refrigeration of the container is necessary. *Category II.* (The proper storage temperature is product-specific and is determined by many factors such as stability of ingredients and optimal activity of antibacterial preservatives; bacteria survivial in some con-

BEST DOCUMENT AVAILABLE

tainers may be enhanced by refrigeration *(17)*. Unless an expiration date is stated on the product label or package insert, it is not known if multiple-use containers, once entered, should be discarded after a specific or arbitrary length of time.)

f. A distinctive supplementary label should be attached to each admixed parenteral stating, as a minimum, the additives and their dosage, the date and time of compounding, the expiration time, and the person who did the compounding. *Category I*

g. All admixed fluids should be refrigerated or started within 6 hours of admixing.
Category II

h. If necessary, admixed parenterals may be stored in the refrigerator for up to a week before use provided refrigeration is continuous and begins immediately after admixing. Other factors, such as stability of ingredients, may dictate a shorter storage time.
Category II (This recommendation is intended to prevent waste of parenterals that are admixed for immediate use but, unexpectedly, cannot be used.)

i. Once started, all parenterals should be completely used or discarded within 24 hours.
Category I

j. Infusions of lipid emulsions should be completed within 12 hours of starting.
Category I

14. IV Filters

Using IV in-line filters is not recommended as a routine infection control measure.
Category Ii

15. IV Teams

Using professional, specially trained IV teams that insert and maintain IV cannulas may decrease the risk of IV-related infections.
Category III

REFERENCES

1. Rhame FS, Maki DG, Bennett JV. Intravenous cannula-associated infections. In: Bennett JV, Brachman PS, eds. Hospital Infections. Boston: Little, Brown and Company, 1979:433-42.

2. Maki DG, Weise CE, Sarafin HW. A semiquantitative culture method for identifying intravenous-catheter-related infection. N Engl J Med 1977;296:1305-9.

3. Band JD, Alvarado CJ, Maki DG. A semiquantitative culture technic for identifying infection due to steel needles used for intravenous therapy. Am J Clin Pathol 1979;72:980-4.

4. Maki DG. Epidemic nosocomial bacteremias. In: Wenzel RP,ed. Handbook of hospital-acquired infection. Boca Raton, Florida: CRC Press, Inc. 1981:371-512.

5. Goldmann DA, Maki DG, Bennett JV. Intravenous infusion-associated infections. In: Bennett JV, Brachman PS, eds. Hospital Infections. Boston: Little, Brown and Company, 1979:443-52.

6. Goldmann DA, Martin WT, Worthington JW. Growth of bacteria and fungi in total parenteral nutrition solutions. Am J Surg 1973;126:314-8.

7. Melly MA, Meng HC, Schaffner W. Microbial growth in lipid emulsions used in parenteral nutrition. Arch Surg 1975;110:1479-81.

8. Tully JL, Friedland GH, Goldmann DA. Complications of intravenous therapy with steel needles and small-bore teflon catheters. Am J Med 1981;70:702-6.

9. Moran JM, Atwood RP, Rowe MI. A clinical and bacteriologic study of infections associated with venous cutdowns. N Engl J Med 1965;272:554-9.

10. Maki DG, Band JD. Study of polyantibiotic and iodophor ointments in prevention of vascular catheter-related infection. Am J Med 1981;70:739-44.

11. Maki DG, MacCormick KN. Acetone "defatting" in cutaneous antisepsis. Crit Care Med 1981;9:202.

12. Dudrick SJ, Diller BG, Wilmore DW. Long term venous catheterization in infants. Surg Gynecol Obstet 1969;129:805-8.

13. von Meyenfeldt M, Stapert J, Wesdorp R, et al. Subclavian catheters: to tunnel or not to tunnel? J Parenter Enter Nutr 1979;3:511.

14. Buxton AE, Highsmith AK, Garner JS, Stamm WE, Dixon RE, McGowan JE. Contamination of intravenous infusion fluid: Effects of changing administration sets. Ann Intern Med 1979;90:764-8.

15. Band JD, Maki DG. Safety of changing intravenous delivery systems at longer than 24-hour intervals. Ann Intern Med 1979;91:173-8.

16. Maki DG, Goldmann DA, Rhame FS. Infection control in intravenous therapy. Ann Intern Med 1973;79:867-87.

17. Lehmann CR. Effect of refrigeration on bactericidal activity of four preserved multiple-dose injectable drug products. Am J Hosp Pharm 1977;34:1196-1200.

BEST DOCUMENT AVAILABLE

Guideline for Prevention of Infections Related to Intravascular Pressure-monitoring Systems

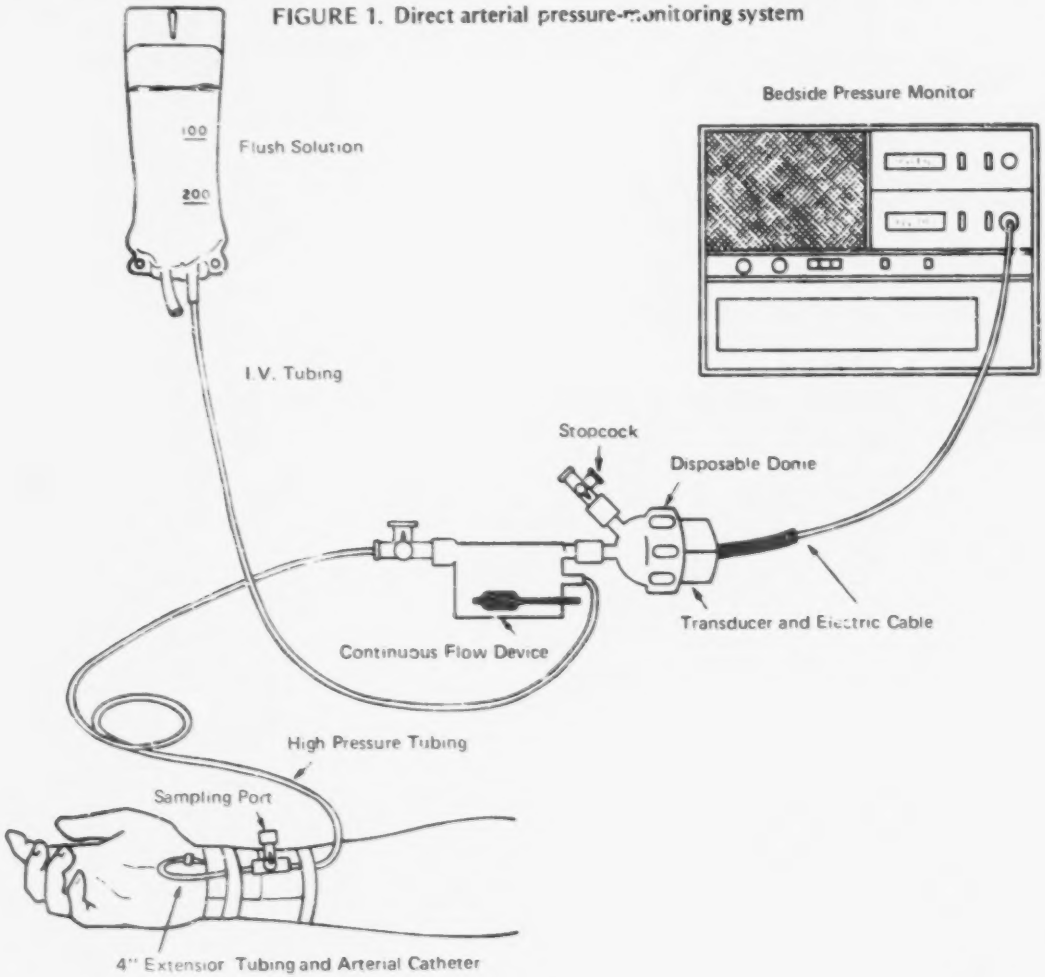
INTRODUCTION

Invasive pressure-monitoring systems are widely used in hospitals throughout the United States, usually for monitoring critically ill patients. These systems have many clinical applications, including monitoring the following pressures: peripheral arterial, pulmonary wedge (through Swan-Ganz catheters), heart chamber, umbilical artery, intrauterine, and intracranial. This guideline, however, will primarily deal with electronic systems that are used to monitor intravascular pressures.

An electronic, direct pressure-monitoring system consists of a fluid-filled cannula (catheter) with one end designed to be placed in the intravascular space (or body cavity) for which the fluid pressure is to be measured and with the other end designed to be connected to an electronic pressure transducer (Figure 1). The transducer converts fluid pressure into electric signals that can be displayed on a monitor. To assure accurate pressure measurement, the system must periodically be calibrated and the cannula and tubing must be kept free of blood clots and other obstructions that prevent free transmission of pressure waves. Thus, the cannula and tubing must be flushed to prevent blood clots from forming at the tip of the cannula. Flushing can be continuous, using a device to deliver a constant flow of several milliliters per hour of flush solution, or intermittent, using a syringe and a stopcock, although the latter method usually requires invasion of a closed system.

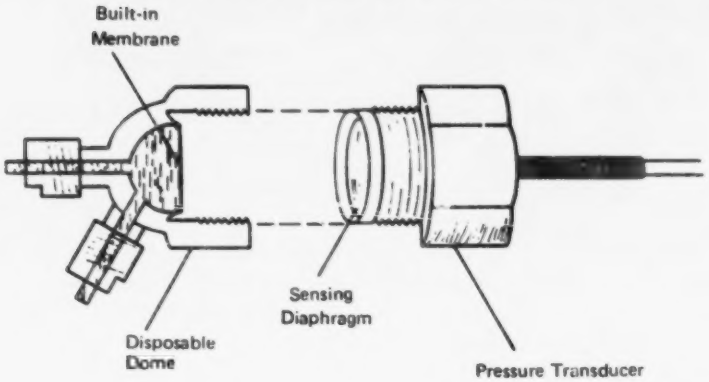
A variety of transducers and other components for pressure-monitoring systems are available for use. Some of these are designed to withstand reprocessing, while others are disposable.

FIGURE 1. Direct arterial pressure-monitoring system



BEST DOCUMENT AVAILABLE

FIGURE 2. Parts of disposable dome transducer



EPIDEMIOLOGY

Many complications of invasive pressure monitoring have been described in the medical literature *(1-6)*. Infectious complications have resulted in significant morbidity and even death in the patients being monitored *(1-5,7-15)*. Because these complications might be preventable with appropriate care of the equipment and patient, special attention must be given to using correct techniques *(1,10,11)*.

Intravascular pressure monitoring is probably the most commonly used type of invasive pressure monitoring. Bacteremia is the most frequent infectious complication reported in association with intravascular pressure monitoring *(1,7-9,11)*. Bacteremia can occur either from contamination of fluid and monitoring components external to the skin or from infection of the cannula insertion site (cannula-related bacteremia) *(11)*. Most epidemics of bacteremia appear to be caused by hospital-acquired contamination of fluid and components external to the skin, such as transducer heads or domes; the importance of external components in infections is supported by the finding that replacing the disposable dome and delivery system every 48 hours virtually eliminates the risk of fluid contamination and related bacteremia *(1)*.

In initial reports of epidemics related to intravascular monitoring, bacteremia was reported to have occurred in patients monitored with a system that had a reusable dome as an integral part of the transducer. In these epidemics, microorganisms causing bacteremia were isolated from the heads of reuse transducers. Once contaminated, these transducers apparently acted as a reservoir of microorganisms because the transducers received inadequate cleaning and disinfection between patient uses *(7,8)*. These epidemics abruptly ceased after measures were taken to assure adequate reprocessing and to prevent recontamination of transducer heads and domes.

Disposable domes were designed to reduce the risk of infections related to pressure monitoring by incorporating a thin membrane to separate the fluid space in contact with the vascular system from any microorganisms that contaminate the transducer head *(9)* (Figure 2). Several outbreaks investigated since 1975, however, have occurred in patients monitored with transducers fitted with disposable domes *(9,15)*. Contamination of the interspace between the transducer and the protective dome membrane has occurred consistently in these outbreaks, but similar contamination of stopcocks and sampling ports in the system has also been documented. It is believed that contamination of the exterior of the system, perhaps at assembly *(15)*, results in contamination of flush fluid and then bacteremia, but the precise mechanism remains undocumented.

In addition to bacteremia associated with the flush fluid, localized infection at the site of cannula insertion also occurs. With arterial cannulas, this is more common with insertion by surgical cutdown than by percutaneous puncture *(11)*. The frequency of infection related to arterial pressure monitoring, both local and systemic, increases significantly if the cannula is left in place for more than 4 days *(11)*.

Infectious complications have also been reported following monitoring of intracranial pressure and intrauterine pressure during labor *(4,6,12-14)*. Although some infection control recommendations for systems used to monitor these pressures may be similar to those used to monitor intravascular pressure, the following recommendations are intended for the latter.

RECOMMENDATIONS

1. Indications for Use

Invasive pressure monitoring should be used only in clinical situations in which information gathered by this technique can clearly influence decisions in patient management. *Category I*

2. Assembly

a. Disposable components that are preassembled and sterile-packaged by the manufacturer should be used when possible. *Category II*

b. Pressure-monitoring systems should be assembled in the simplest arrangement possible. Sterile items, including disposable domes and lines, should be kept in their sterile wrapping until needed. These items and the transducer should not be assembled hours or days before the time of actual need even if this is to prepare for a possible emergency. Most important, systems should never be filled with flush solution and stored (since even a few microorganisms inadvertently introduced into the solution at the time of assembly can rapidly multiply during storage). *Category I*

c. In disposable domes and transducers, the space between the transducer head and dome membrane should be left dry or, if fluid is required, the space should be filled with normal saline, bacteriostatic water, or 70% alcohol (if the manufacturer states that it will not damage the dome or transducer); glucose-containing solutions should not be used since they are known to support the growth of many microorganisms. *Category I*

3. Flushing Lines

a. Patency of pressure-monitoring cannulas should be maintained through the use of a closed flush system rather than an open system that requires use of a syringe and stopcock. *Category II*

b. Flush solutions, i.e., those whose main purpose is to maintain patency of the cannula and which infuse very slowly or are given as intermittent boluses, should not contain glucose for the reasons stated above. *Category I*

4. Handwashing

a. Hospital personnel should wash their hands before inserting a pressure-monitoring cannula or manipulating a pressure-monitoring system, such as to draw a blood specimen. *Category I*

b. An antiseptic handwashing agent is preferred, but soap and water can be used. *Category II*

c. 1) Sterile gloves should be worn for insertion of central cannulas for pressure monitoring (for example, pulmonary artery and central venous cannulas) and for peripheral cannulas requiring a cutdown. *Category I*

2) Sterile gloves should be worn for insertion of other intravascular cannulas used for pressure monitoring. *Category III*

5. Insertion and Maintenance

Recommendations for insertion and maintenance of *intravascular pressure-monitoring systems* are similar to those recommended for *intravenous cannulas.*

a. The site chosen for cannula insertion should be scrubbed with an antiseptic prior to insertion of the cannula. *Category I*

b. Tincture of iodine (1%-2%) is preferred for the insertion site, but chlorhexidine, iodophors, or 70% alcohol can be used. The antiseptic should be applied liberally and allowed to remain in contact for at least 30 seconds before insertion. *Category II*

c. Neither aqueous benzalkonium-like compounds nor hexachlorophene should be used to scrub the insertion site. *Category I*

d. A topical antibiotic or antiseptic ointment should be applied at the insertion site immediately after cannula insertion especially for insertions by cutdown. *Category II*

e. Central and cutdown cannulas should be inserted with aseptic technique and sterile equipment. Gloves and drapes are usually required to achieve this objective. *Category I*

f. The cannula should be secured to stabilize it at the insertion site. *Category I*

g. A sterile dressing should be applied to cover the insertion site. The dressing, and not tape, should cover the wound unless the tape is sterile. *Category I*

h. The date of insertion should be recorded in a place where it can be easily found. *Category I* (The date may be recorded in the medical record and, if feasible, on the dressing or tape.)

i. Patients with intravascular devices should be evaluated at least daily for evidence of cannula-related complications. This evaluation should include gentle palpation of the insertion site through the intact dressing. If the patient has an unexplained fever or there is pain or tenderness at the insertion site, the dressing should be removed and the site inspected. *Category I*

j. For peripheral cannulas that must remain in place for prolonged periods, the insertion site should be inspected and dressed with a new sterile dressing at 48-72 hours. Thereafter, the site should be inspected and dressed regularly (the optimal frequency of dressing cannula sites in this situation is not known). *Category II*

k. For central cannulas that must remain in place

BEST DOCUMENT AVAILABLE

for prolonged periods, the insertion site should be inspected and dressed with a new sterile dressing every 48-72 hours. *Category II*

l. Antibiotic or antiseptic ointment, if used, should be reapplied with each dressing change. *Category II*

6. Calibration

During calibration of a pressure-monitoring system, contact should not occur between the sterile fluid column in the cannula and tubing and nonsterile solutions or equipment. *Category I*

7. Obtaining Specimens

a. Ideally, pressure-monitoring systems should be maintained as closed systems. Stopcocks, if used, should be covered. *Category I*

b. Arterial pressure-monitoring systems should be used primarily to monitor blood pressure and to obtain specimens for arterial blood-gas analysis. Routine blood specimens, if they are to be drawn from the arterial line, should be drawn at the same time as those for blood-gas analysis, if possible, to keep the number of manipulations to a minimum. *Category I*

c. Care should be taken to ensure that all specimens be obtained aseptically; for example, syringes chilled in ice should not be allowed to contaminate the stopcocks or sampling ports *(7). Category I*

8. Replacing Intravasular Monitoring Systems in Patients Requiring Prolonged Monitoring

a. The container of flush solution should be changed every 24 hours. *Category I*

b. The chamber dome, administration tubing, and continuous flow device (if used) should be replaced at 48-hour intervals. *Category II* (It is not known whether the transducer needs periodic disinfection or sterilization during prolonged use on a single patient.)

c. Under special circumstances, such as following the reflux of blood into the tubing or domes or following countershock (which could damage the protective membrane of some disposable domes), replacement of the tubing and dome is desirable. *Category III*

d. Peripheral arterial cannulas should be left in place no longer than 4 days *(11)* if other sites for cannula insertion are available. *Category II*

e. Central cannulas for pressure monitoring that are inserted through a *peripheral vein* should be removed in 48-72 hours. *Category I* (These cannulas may occasionally have to be used for longer than 48-72 hours if another insertion site cannot be found. The proper frequency for changing pressure-monitoring cannulas inserted through a subclavian or jugular approach is not known).

f. Cannulas for pressure monitoring should not be replaced over a guide wire if this is done solely for infection control. *Category II*

g. Intravascular pressure-monitoring systems should be removed when they are no longer medically indicated and promptly discontinued or placed at another site if the initial site becomes infected or if the monitoring system is suspected as the source of clinical sepsis or bacteremia. *Category I*

9. Processing Transducers Before Reuse

a. Disposable components of the pressure-monitoring system should not be resterilized and reused. *Category I*

b. 1) After use, transducers (including transducer heads and reusable domes) should be cleaned, disinfected (high-level) with a chemical agent or sterilized with ethylene oxide, and stored in a manner to prevent recontamination before use on the next patient. *Category I* (The manufacturers of the transducer and reusable dome should be asked for recommendations about classes of disinfectants that will not injure the equipment and about proper methods of cleaning, sterilizing, and storing components.)

2) Sterilization with ethylene oxide is recommended unless the manufacturer states that this method is not satisfactory. *Category II*

BEST DOCUMENT AVAILABLE

REFERENCES

1. Maki DG, Hassemer CA. Endemic rate of fluid contamination and related septicemia in arterial pressure monitoring. Am J Med 1981;70:733-8.

2. Emergency Care Research Institute: Evaluation, blood pressure monitors. Health Dev 1975;5:8-22.

3. Lantiegne KC, Civetta JM. A system for maintaining invasive pressure monitoring. Heart Lung 1978;7:610-21.

4. McDowall DG: Monitoring the brain. Anesthes 1976;45:117-34.

5. Finster M, Petrie RH. Monitoring of the fetus. Anesthes 1976; 45:198-215.

6. Banta HD, Thacker SB. Costs and benefits of electronic fetal monitoring: review of the literature. National Center for Health Services Research, 1979. (DHEW publication no. [PHS] 79-3245).

7. Stamm WE, Colella JJ, Anderson RL, Dixon RE. Indwelling arterial catheters as a source of nosocomial bacteremia. N Engl J Med 1975;292:1099-1102.

8. Weinstein RA, Stamm WE, Kramer L, Corey L. Pressure monitoring devices—overlooked source of nosocomial infection. JAMA 1976;236:936-8.

9. Buxton AE, Anderson RL, Klimek J, Quintiliani R. Failure of disposable domes to prevent septicemia acquired from contaminated pressure transducers. Chest 1978;74:508-13.

10. Retailliau HF. Infection control with invasive pressure monitoring devices. AFIC Journal 1979;7:13-7.

11. Band JD, Maki DG. Infections caused by arterial catheters used for hemodynamic monitoring. Am J Med 1979;67:735-41.

12. Gibbs R, Jones PM, Wilder CJY. Internal fetal monitoring and maternal infection following cesarean section--prospective study. Obstet Gynecol 1978;52:193-7.

13. Gassner CB, Ledger WJ. The relationship of hospital-acquired maternal infection to invasive intrapartum monitoring techniques. Am J Obstet Gynecol 1976;126:33-7.

14. Hagen D. Maternal febrile morbidity associated with fetal monitoring and cesarean section. Obstet Gynecol 1975;46:260-2.

15. Donowitz LG, Marsik FJ, Hoyt JW, Wenzel RP. *Serratia marcescens* bacteremia from contaminated pressure transducers. JAMA 1979;242:1749-51.

U.S. GOVERNMENT PRINTING OFFICE: 1982-360-997/2090